A Report to the Utah Division of Water Quality

Ecological analyses of nutrients, plankton and benthic communities in Farmington Bay and the Great Salt Lake, Utah (2004)

Aquatic Ecology Practicum Class Project 2004
Department of Aquatic, Watershed and Earth Resources
Utah State University

Amy Marcarelli, Teaching Assistant
Wayne Wurtsbaugh, Instructor[1]

Students

Jessica Horrocks
Robert S. Jensen
Kathleen B. Markland
Jonas N. Parker
Justin Robinson
Erin Van Dyke

February 14, 2005

A Report to the Utah Division of Water Quality

Ecological analyses of nutrients, plankton and benthic communities in Farmington Bay and the Great Salt Lake, Utah (2004)

Aquatic Ecology Practicum Class Project 2004
Department of Aquatic, Watershed and Earth Resources
Utah State University

Amy Marcarelli, Teaching Assistant
Wayne Wurtsbaugh, Instructor[1]

Students

Jessica Horrocks
Robert S. Jensen
Kathleen B. Markland
Jonas N. Parker
Justin Robinson
Erin Van Dyke

February 14, 2005

Reference:
Marcarelli, A.M., W.A. Wurtsbaugh, J. Horrocks, R.S. Jensen, K.B. Markland, J.N. Parker, J. Robinson and E. VanDyke. 2005. Ecological analyses of nutrients, plankton and benthic communities in Farmington Bay and the Great Salt Lake, Utah (2004). Aquatic Ecology Practicum (AWER 4510) Class Project. College of Natural Resources, Utah State University. 71 p.

[1]Author for correspondence: Dept. Aquatic, Watershed and Earth Resources, Utah State University, Logan, Utah 84322-5210. 435 797-2584, wurts@cc.usu.edu

Table of Contents

Executive summary 3

Chapter 1. Nutrient loading to the Great Salt Lake from sewage, riverine and atmospheric sources 6
ROBERT S. JENSEN

Chapter 2. Nitrogen fixation across a salinity gradient: death by salt...a true story 16
JUSTIN ROBINSON

Chapter 3. Ecology of stromatolitic structures in the Great Salt Lake, Utah 26
JONAS N. PARKER

Chapter 4. Cryptobiology, paleolimnology and selenium sensitivity of brine shrimp from the Great Salt Lake, Utah, USA 39
KATHLEEN B. MARKLAND

Chapter 5. Survival of *Artemia* in the Great Salt Lake, Utah 49
ERIN VANDYKE

Chapter 6. Predation by *Trichocorixa verticalis* on *Artemia franciscana*: effect of light on selection of prey 63
JESSICA HORROCKS

Executive Summary

In Fall 2004, the Aquatic Ecology Practicum class at Utah State University finished a third year of research on limnological and ecological characteristics of Farmington Bay and Gilbert Bays of the Great Salt Lake. Our previous research has produced interesting findings in Farmington Bay, including hypereutrophy (Marcarelli et al. 2001), high phosphorus loading into the Bay, overnight water column anoxia linked to high winds (Wurtsbaugh et al. 2002), potential predator control of brine shrimp, and high levels of hydrogen sulfide in the sediment and deep brine layer (Marcarelli et al. 2003). These class findings have lead to increased interest in Farmington and Gilbert Bays. Because of the breadth of research now occurring in Farmington Bay, the topics studied by the students this fall encompassed a wider range of research than ever before. The reports ranged from an expanded analysis of nutrients entering Great Salt Lake, including external loading and biological nitrogen fixation, benthic ecology of Gilbert Bay including analyses of stromatolites and brine shrimp cysts in sediments, and more focused experiments on brine shrimp survival and predation by corixids in Farmington Bay. Key findings of the students are identified below.

Nitrogen Loading and Nitrogen Fixation in Gilbert and Farmington Bays

A nutrient budget constructed in 2001 identified high phosphorus loading into Farmington Bay from surrounding sewage treatment plants, with loads 8-times greater than necessary to cause the bay to be hypereutrophic (Wurtsbaugh et al. 2002). This year, Robert Jensen (Ch. 1) assessed nutrient loading to the entire Great Salt Lake, as well as Farmington Bay, using data available in STORET. His analysis showed that loading rates to both bays were greater than the critical loading level to cause eutrophication, but loads into Farmington Bay were 10 times greater than into Gilbert Bay. Discharge from Farmington Bay contributed 21% of the load to Gilbert Bay and atmospheric loading contributed an additional 15%. In Farmington Bay, 43% of the phosphorus loading came from sewage treatments plants in Davis and Salt Lake Counties.

One source of nitrogen for algal communities is nitrogen fixation by pelagic cyanobacteria, which is controlled in Great Salt Lake by nutrient supply and salinity levels (Wurtsbaugh and Marcarelli 2004a). This fall, low salinity levels (3%) in Farmington Bay led to a bloom of the nitrogen fixing cyanobacteria *Nodularia* sp., allowing an examination of the distribution of these algae in Farmington and Gilbert Bays (Justin Robinson, Ch. 2). Algal levels (Chl. a, 125 $\mu g\ L^{-1}$) and nitrogen fixation was very high in Farmington Bay, but quickly fell off as water moved through the causeway breach into Gilbert Bay where salinity was much greater. A laboratory experiment confirmed that *Nodularia* exposed to salinities above 10% ceased nitrogen fixation in less than 4 hours, indicating a rapid physiological response of these cells to increased salinity. Finally, isotope analysis suggested that nitrogen fixation may supply a significant amount of nitrogen to plankton in Farmington Bay when conditions are favorable.

Benthic Ecology and Cryptobiology of Gilbert Bay

The benthic communities of Great Salt Lake have rarely been studied. One important component of the lake bottom is stromatolites in the shallow sections of Gilbert, and perhaps Farmington Bay. Jonas Parker (Ch. 3) made a series of field and laboratory measurements to characterize stromatolites in two areas near Antelope Island. Stromatolites in the lake are formed as a union between carbonate precipitates and a near uniculture of the cyanobacteria *Aphanothece* sp. A laboratory bioassay was unable to detect nutrient limitation of the community and indicated that there was no nitrogen fixation occurring on the stromatolites. However, there is much work to be done to understand how these structures fit into the overall Great Salt Lake food web.

Much of the bottom of the Great Salt Lake is underlain by sediments, and each year millions of brine shrimp cysts are buried in these sediments, forming an egg bank in the

sediments of Gilbert Bay. This fall, Kathleen Markland collected sediment cores from Gilbert Bay to determine the presence and viability of cysts in this egg bank (Ch. 4). She successfully hatched cysts from as deep as 25-26 cm, which were estimated to be 360 years old, pre-dating Anglo settlement of the Great Salt Lake valley. These brine shrimp are being maintained in our laboratory and can be used in future experiments to determine whether brine shrimp have evolved resistance mechanisms to pollutants in the lake.

Pelagic Ecology of Farmington and Gilbert Bays

Industry and agencies have recently focused on the effects of selenium (Se) on organisms in the Great Salt Lake, as work progresses towards a numeric standard for Se specific to the Lake. A bioassay estimated the 48-hour LC_{50} for newly hatched brine shrimp nauplii to be 27 mg Se L^{-1} (Markland, Ch. 4), which was considerably less than a site specific toxicity level previously estimated for the Great Salt Lake (Brix et al. 2004). These results suggest that more work is needed to determine the toxicity level of selenium for brine shrimp.

Smaller populations of brine shrimp are found in Farmington than in Gilbert Bay during most of the year (Wurtsbaugh et al. 2002, Wurtsbaugh and Marcarelli 2004b), and the factors driving this difference remain unclear. Erin VanDyke (Ch. 5) conducted in situ cage and laboratory experiments to assess the importance of water quality for brine shrimp survival in the two bays. Despite problems with the cage construction, field results showed that conditions at 0.8-m in Farmington Bay lead to mortality of all zooplankton. A laboratory assay confirmed this result, with no survival of brine shrimp nauplii in either deep Farmington Bay or surface Gilbert Bay water. In Gilbert Bay, it was likely that food in the surface water was too low to support brine shrimp. In Farmington Bay, high hydrogen sulfide concentrations (20 mg L^{-1}) and anoxia at 0.8-m were likely responsible for zooplankton mortality, as has been suggested previously (Wurtsbaugh and Marcarelli 2004c).

Another potential control of brine shrimp in Farmington Bay is predation by the water boatman, *Trichocorixa verticalis* (Marcarelli et al. 2003, Marcarelli and Wurtsbaugh 2004). Previous experiments have shown that corixids can feed more effectively on juvenile brine shrimp than on adults or nauplii, leading to the question of whether corixids are visual or tactile predators. To answer this question, Jessica Horrocks (Ch. 6) conducted laboratory predation experiments with light and dark treatments to determine how corixids locate their prey. In dark treatments, corixids had moderately lower predation rates on nauplii compared to predation rates on larger brine shrimp, suggesting that the increased movements of the adult shrimp might facilitate tactile predation. In the light treatments corixids preyed more heavily on nauplii than adults, suggesting that under high light conditions corixids can use visual cues to locate prey. These results, however, were not conclusive and additional laboratory and field experiments are needed to determine if corixids are truly tactile and visual predators.

References

Brix, KV, DK Deforest, RD Cardwell and WJ Adams. 2004. Derivation of a chronic site-specific water quality standard for selenium in the Great Salt Lake, Utah, USA. Environmental Toxicology and Chemistry 23: 606-612.

Marcarelli, AM, MD Mills, and WA Wurtsbaugh. 2001. The Great Salt Lake doesn't stink...but Farmington Bay does! Friends of Great Salt Lake Newsletter 7(2): 5, 15; 7(4): 6.

Marcarelli, AM, WA Wurtsbaugh, B Albrecht, E Archer, J Bassett, M Beckstrand, M Hadley, J Kling, O Lester, P MacKinnon and T-h Ting. 2003. Continuing studies of water quality in Farmington Bay and the Great Salt Lake, Utah. Aquatic Ecology Practicum Class Project 2002. College of Natural Resources, Utah State University. 91 p.

Marcarelli, AM and WA Wurtsbaugh. 2004 (abstract). Predation by an invertebrate predatory in Farmington Bay, Great Salt Lake, Utah. Utah State University Runoff Conference, Logan, UT.

Wurtsbaugh, WA, AM Marcarelli, D Gross, J Moore, C Christison, S Kircher and S Bates. 2002. Comparative analysis of pollution in Farmington Bay and the Great Salt Lake, Utah. Aquatic

Ecology Practicum Class Project 2001. College of Natural Resources, Utah State University. 21p.

Wurtsbaugh, WA and AM Marcarelli. 2004a. Analysis of phytoplankton nutrient limitation in Farmington Bay and the Great Salt Lake. Report to the Central Davis Sewage Improvement District. 57 p.

Wurtsbaugh, WA and AM Marcarelli. 2004b. Phytoplankton and zooplankton in Farmington Bay and the Great Salt Lake, Utah (2003). Report to the Central Davis Sewage Improvement District, Kaysville, UT. 57 p.

Wurtsbaugh, WA and AM Marcarelli. 2004c. Hydrogen sulfide in Farmington Bay and the Great Salt Lake: a potential odor-causing agent. Report to the Central Davis Sewage Improvement District, Kaysville, UT. 30 p.

Chapter 1

Nutrient Loading to the Great Salt Lake from Sewage, Riverine and Atmospheric Sources

Summary

Nutrient loading to the Great Salt Lake is a concern because it may cause eutrophication and degrade water quality. The amount of phosphorus entering the lake and its wetlands from tributaries and point sources was estimated using data collected by the Utah Division of Water Quality and the USGS available on the STORET database. Atmospheric loading was estimated using data adapted from studies preformed in the Mediterranean Sea. In 1999-2000 estimated areal loading of Farmington Bay was 2.99 g P m^{-2} y^{-1}, and loading to Gilbert Bay was 0.26 g P m^{-2} y^{-1}; both loading levels are well above the critical loading level of 0.13 g P m^{-2} y^{-1} thought to cause eutrophic conditions in lakes. Davis County sewage treatment plants and the Sewage Canal from Salt Lake City contribute 43% of the loading to Farmington Bay. Although loading rates to both bays are high, an unknown proportion of some of the sources is removed by wetlands prior to reaching the lake. More than 21% of the water entering Farmington Bay was from sewage treatment plants. Dominant phosphorus sources for Gilbert Bay were the Bear River (34%), Goggins Drain (24%), Farmington Bay outflow (21%) and atmospheric deposition (15%). Because the Great Salt Lake and most other saline systems are limited by nitrogen rather than phosphorus we used N:P ratios collected from riverine and sewage effluent in October 2004 to estimate nitrogen loading to the lake. Nitrogen loading rates were also very high to Gilbert Bay and particularly Farmington Bay. Removing the automobile and railway causeways would improve loading rates into Farmington Bay by increasing dilution, but overall loading rates to the open lake would still be above critical levels. Because nutrient loading rates to the Great Salt Lake are already high and population growth in this closed basin is increasing, a more thorough analysis of nutrient loading to the lake and impacts on the biota are needed.

Robert S. Jensen
Department of Aquatic Watershed and Earth Resources
Utah State University
Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 Dec 2004

Introduction

Anthropogenic increases in nutrient loading to lakes may cause eutrophication and consequently decrease water quality and habitat available for aquatic organisms (Mason 2002). Since the Clean Water Act was instituted in 1972 water quality has improved in many lakes as point source pollution has been controlled nationwide. However, pollutant control has not been instituted on all water bodies in the United States, particularly those that are not used as drinking water sources. Utah's Great Salt Lake continues to receive considerable nutrients and other pollutants from both point and non-point sources.

A large amount of the water that enters the Great Salt Lake comes through sewage treatment plants. This water is high in phosphorus and nitrogen that contributes to eutrophication (Mason 2002). Bioassays of Great Salt Lake waters have indicated that the limiting nutrient is nitrogen (Stephens and Gillespie 1976; Wurtsbaugh 1988). However, most lakes are limited by phosphorus (Dodds 2002), and thus data for phosphorus is much more readily available from state agencies that monitor water quality. Because of data availability, phosphorus loading to the Great Salt Lake can be estimated. Nitrogen loads can also be estimated for the Great Salt Lake using an N: P ratio from water samples taken from some of the tributaries and point sources in October (see below), and assumed to comparable for the rest of the year.

In this study, nutrient loading of the Great Salt Lake was estimated for Farmington and Gilbert Bays individually. Phosphorus concentrations from 1 Aug 1999 to 31 July 2000 were analyzed (Data source STORET) for all major tributaries of the Great Salt Lake. In Farmington Bay this includes the Jordan River, the Sewage canal, the Surplus Canal and discharge from four wastewater treatment plants located in Davis County, while in Gilbert Bay this includes Farmington Bay, the Goggins Drain, the Weber River, and the Bear River.

In addition to these budgets, we used our calculations to examine the impacts of anthropogenic activities on the nutrient loads to the lake. For example, if the causeways that divide the lake into its three sections were to be removed, the areal loading to the different sections of the lake would also change. Consequently, estimated loads for this scenario were also determined. Another interesting value that was determined was the amount of water entering Farmington Bay from sewage treatment plants in Davis County vs. the amount entering from more natural sources, such as Jordan River and the Sewage Canal. However, a large portion of the phosphorus in the "more natural" Jordan River is from wastewater treatment plants located in Salt Lake County (Loving et al. 1988), so this value should be considered with some caution.

Study Area and Methods

Study Area–The Great Salt Lake can is separated into three parts. Farmington Bay is located in the southeast corner of the lake; it has an area of 259 km^2 and is separated from Gilbert Bay by a road causeway providing access to Antelope Island. Because of the large nutrient loads entering Farmington Bay it has become the most eutrophic lake in Utah (Wurtsbaugh et al. 2002). Within the causeway there is a small breach allowing water from Farmington Bay to flow into Gilbert Bay and vice versa. Two culverts also allow water to exchange between the bays, but these are sometimes dry. Gilbert Bay is considered the main body of the lake and has an area of 2366 km^2. The north part of the lake, Gunnison Bay, has an area of 1559 km^2. It is separated from Gilbert Bay by a railroad causeway and there is also limited water exchange confined to three breaches, culverts and percolation through the earth and rock causeway (Loving et al. 1988).

Personal Water Sampling Sites–During the second week of October I sampled some of the tributaries of the Great Sale Lake. A Bear River sample was collected just northwest of Brigham City where 900 North crosses the river (41.5459°N, 112.0959°W). A Jordan River sample was collected at the Cudahy Lane crossing (40.8421°N, 111.9513°W). A Sewage Canal sample was also collected at the Cudahy Lane crossing (40.8421°N, 111.9537°W). A Central Davis Sewage Treatment sample was collected directly where water discharges from the treatment plant (40.9992°N, 111.9462°W). Water samples were collected in 125-ml acid washed plastic bottles. The samples were then delivered to Dr. Michelle Baker's laboratory at Utah State University where Dr. Agnes Chartier performed total phosphorus and nitrogen analyses using a persulfate digestion followed by second derivative analysis for TN and a colorimetric technique using malachite green for TP to determine concentrations ($\mu g\ L^{-1}$) and N:P ratios for tributary waters. This data, however, was part of the collection and analyzing part of the class and data from this collection will not be included in this report.

Data Sources–Data for phosphorus loads and flows for all tributaries were obtained from the EPA's STORET database (http://www.epa.gov/storet) for the period from 1 August 1999 to 21 July 2000. The Division of Water Quality ID numbers used to access data on STORET were:

Bear River # 4901100
Weber River # 4920050
Jordan River # 449182
Sewage Canal # 4991050
Central Davis Sewage Treatment Plant # 499027
North Davis Sewage Treatment Plant # 499007
South Davis North Sewage Treatment Plant # 499078
South Davis South Sewage Treatment Plant # 499181

In addition, data for the Surplus canal (# 4991310) were obtained from the DWQ database maintained by A. Hultquist. Nutrient data for the Goggins Drain were not available. Consequently, we estimated loading using phosphorus loads from the Surplus Canal and discharges from a USGS published water balance report (Loving et al. 1988). This approach was reasonable because Goggins Drain is fed from the Surplus Canal and therefore should have very similar nutrient concentrations.

Atmospheric phosphorus loading was based on the data of Dorm et al. (2004), who estimated that the deposition of bioavailable P to the Mediterranean Sea was 19.5 mg $m^{-2}\ yr^{-1}$. Bioavailable P is approximately 50% of total P in dust (Herut et al. 2002). Consequently, Dorm's bioavailable P estimates were doubled because all of the other source data used for the budgets were based on total phosphorus concentrations. Dust inputs to the Mediterranean are high because of the proximity of the Saharan Desert. Dust sources from Utah's West Desert were assumed to be comparable to those of the Sahara. The assumed deposition rate of 39 mg $m^{-2}\ yr^{-1}$ is within the upper range of deposition rates noted by Redfield (2002) in a review of atmospheric P deposition.

Data Analysis Methods to Determine TP and TN Loading–Phosphorus loads were estimated by multiplying concentrations on a given date by discharges on that date. These daily load estimates were then multiplied by the number of days between sampling intervals. These estimates were summed to provide an annual load (kg P y^{-1}). In many cases there was more then a month of data missing; in those cases the monthly loading was estimated by interpolation. This process was repeated for all tributary data collected. Phosphorus loading to Gilbert Bay from the outflow of Farmington Bay was determined using the estimated hydraulic residence time of Farmington Bay (0.77 yr^{-1}; Austin 1993). Based on this value, we estimated that 33% (1-0.77) of the water entering the bay was flushed into Gilbert Bay. This volume was

multiplied by the mean TP concentration of Farmington Bay (0.63 mg P L^{-1}; W. Wurtsbaugh unpublished data).

The annual phosphorus loads from the Jordan River, Sewage Canal, Surplus Canal and the four Davis County sewage treatment plants were then divided by the surface area of Farmington Bay (259 km^2 at a lake elevation of 4200 feet) to arrive at an estimated annual areal TP loading (g P m^{-2} y^{-1}). This process was then repeated for the annual phosphorus loads entering Gilbert Bay from the Bear River, Weber River, Goggins Drain and Farmington Bay outflow by divided the loads by the surface area of Gilbert Bay (2366 km^2). This process was again replicated to determine the estimated areal TP loading for Gunnison Bay. Areal loading is then integrated into the equation $TP = L / z(p + q)$ to determine an estimated concentration of total phosphorus (Vollenweider 1971), where TP is the estimated concentration of total phosphorus (mg P m^{-3}), L is the areal loading (g P m^{-2} y^{-1}), z is the mean depth (m), p is the hydraulic flushing rate (0.77 yr^{-1} for Farmington Bay, 0.2 for Gilbert Bay, and 0 for Gunnison Bay), and q is the sedimentation rate coefficient for P estimated by Vollenweider (1971) to be 10 m y^{-1}/z (Cooke et al. 1993). Mean depths for Farmington and Gilbert Bays were assumed to be 0.9 and 5.0 m. The hydraulic flushing rate (5 yr^{-1}) for Gilbert Bay was estimated by dividing the cubic meters of water estimated to enter Gunnison Bay each year by the cubic meters of water in Gilbert Bay. The hydraulic flushing rate for Gunnison Bay was determined to be zero because we estimated that no water entering this part of the lake will leave (Loving et al. 1988).

The estimated TP concentrations if the causeways were removed was calculated in the same way, with the exception that mean depths and areas were changed in the calculations to adjust for the combination of areas. The TP was first calculated as if the road causeway between Farmington and Gilbert Bay was removed but the railroad causeway remained. Then TP was calculated as if the railroad causeway between Gilbert and Gunnison was removed but the road causeway remained. Finally, the TP was calculated as if both causeways were removed. In the calculations it was also assumed that there would be complete mixing of all lake water, which would or would not be the case depending on many factors including water temperature, wind and stratification (Dodds 2002).

To approximate total nitrogen load to the lake, we used our estimates of the TN:TP ratio for the Sewage Canal (7:1 by weight), and the Jordan River (11:1) from the samples collected in October, and assumed they were representative of the whole year. These ratios were then multiplied by the phosphorus load estimates for sewage treatment plants or rivers to approximate nitrogen loading. The 11:1 ratio was also applied to the atmospheric loads and the load moving from Farmington Bay into Gilbert Bay.

"Natural" vs. Wastewater Treatment Flows–Flow rates determined above were used to determine what portion of the water entering Farmington Bay comes directly from sewage treatment plants. The amount of water entering the lake from wastewater treatment plants (Davis Co. sewage and the Sewage Canal) were compared with amounts of water entering from more natural flows (Jordan River and Surplus Canal). However, a portion of the water in the more natural flow is also derived from wastewater treatment plants located in the southern half of Salt Lake County and Utah County, so this result will underestimate the proportion of water due to treatment plants.

Results

Total Phosphorus Loading–Estimated total phosphorus loading to Farmington Bay was extremely high at 775,000 kg P yr^{-1}, or 2.99 g m^{-2} yr^{-1} (Table 1). The largest portion of phosphorus entering Farmington Bay is from the Surplus Canal (42%) because it has the largest flow rate of all the inflows examined. Direct sewage inflows comprised 53% of the P

budget, of which 33% was due to the Davis County treatment plants. The Jordan River contributed 14% and atmospheric deposition was 1% of the load (Figure 1a).

The estimated total phosphorus loading to the much larger Gilbert Bay was somewhat smaller than that entering Farmington Bay at 620,000 kg P yr^{-1}. The largest portion of phosphorus entering Gilbert Bay is from the Bear River (31%) because of its high flow rate, followed by the Goggins Drain (24%), outflow from Farmington Bay (21%), atmospheric deposition (15%), and the Weber River (9%; Figure 1b). In Figure 2 all of the tributaries and sewage inflows are plotted together, showing the relationship between Farmington Bay and Gilbert Bay.

Areal rates of loading, calculated by distributing total loads over the respective areas of each bay, demonstrate that phosphorus loading to Farmington Bay is approximately 10 times higher to Farmington Bay than to Gilbert Bay (Figure 3). Estimated areal loading of Farmington Bay was 2.99 g P m^{-2} yr^{-1}, and load to Gilbert Bay was 0.26 g P m^{-2} yr^{-1}. Loading to Farmington Bay far exceeds permissible loading levels for P determined by Vollenweider (1971), where loading for a lake with an average depth of 5 m is permissible up to 0.07 g m^{-2}, (Table 2) but should not exceed 0.13 g m^{-2} yr^{-1} to prevent eutrophication (Figure 3). Furthermore if the causeways were to be removed and full mixing was to occur the areal loading would still be well above the danger zone with 0.48 g m^{-2} yr^{-1} if Gilbert and Farmington Bay were connected, and 0.30 g m^{-2} yr^{-1} if the entire lake was reconnected (Figure 4).

Estimated Nitrogen–Farmington Bay had an estimated nitrogen load of 28 g N m^{-2} yr^{-1} and Gilbert Bay had a load of 2.9 g N m^{-2} yr^{-1}. If the automobile causeway was to be removed to allow nitrogen to mix between these two bays, the estimated loading to the combined area would be 4.8 g N m^{-2} yr^{-1}. These loading levels are also above the permissible loading levels determined by Vollenweider (1971). Permissible levels of nitrogen loading are up to 1.0 g m^{-2}, and dangerous levels are anything above 2.0 g m^{-2} (Table 3).

"Natural" vs. Wastewater Treatment Flows–When water amounts from natural flows were plotted against sewage treatment flows (Figure 5) it showed that 21% of water entering Farmington bay is coming directly from sewage treatment plants, while 79% is entering from more natural flows. During the summer when river flows decrease, treated sewage effluents represent up to 30% of the inflow water. Interestingly, 21% of water entering Farmington Bay from Sewage Treatment Plants contains 44% of the total phosphorus, while the greater natural flow contains only 56% of the total phosphorus. This shows that the smaller flows from the sewage are more highly loaded with nutrients for their size than the natural flows and likely contribute more heavily to eutrophication in the Bay.

Discussion

The estimated phosphorus and nitrogen loading levels indicate that the Great Salt Lake should be highly eutrophic. While many of the numbers used to calculate the P values were averages, leaving room for error, the levels of nutrients entering the lake are so high that even if the true values were only half of what is reported the lake would still be eutrophic. Furthermore, many of the tributaries run through wetlands before entering the lake. These wetlands would remove a significant amount of nutrients so that the true loading rates are likely not as high as reported here. However, even if the wetlands removed 50% of the nutrients that passed through them, nutrient loading to Gilbert, and particularly Farmington Bay would still be high.

Limnological conditions in Farmington Bay support the conclusion that it is hypereutrophic, with chlorophyll concentrations usually well above 100 µg L^{-1} and Secchi depths less than 0.3 m (Wurtsbaugh and Marcarelli 2004a). In contrast, chlorophyll levels in Gilbert Bay are often far less than would be predicted by phosphorus or nitrogen loading and are less

than 5 μg L^{-1} during the summer. This is likely because the abundant brine shrimp are such good grazers that they exert top-down control of the phytoplankton populations (Wurtsbaugh 1992). However, when brine shrimp disappear in the winter, chlorophyll levels in Gilbert Bay rise to over 50 μg L^{-1}, which is a level indicative of a moderately eutrophic state (Wurtsbaugh and Marcarelli 2004a). These levels are still considerably less than those in Farmington Bay, consistent with the 10-fold higher nutrient loading in Farmington than in Gilbert Bay.

Although causeway removal would decrease nutrient levels in Farmington Bay, the large loading of nutrients from this bay that would mix into the larger lake could cause problems in Gilbert and Gunnison Bays. Estimated loading to Gilbert Bay would increase approximately 70%. The impact of this increased loading is hard to assess, given the importance of brine shrimp grazing in controlling phytoplankton.

Although this effort to understand the nutrient budget of the Great Salt Lake and its bays provides some insights about water quality and possible management strategies to control eutrophication, much more detailed efforts will be needed to accurately measure some of the factors addressed in this report. This report was meant only to serve as an estimation to get an idea of the large amounts of nutrients entering the lake from sewage and natural flows. An accurate nutrient budget would require determining the amounts of nutrient absorbed in wetlands and nitrogen loads of all tributaries, including Goggins Drain. Currently much of this data does not exist and information was pooled from many different sources in order to make calculations. However, even though the loading estimates may sway in this report from the true levels of nutrients in the lake, it is apparent from our estimates that the Great Salt Lake is highly eutrophic. Future efforts to relate nutrient loading to eutrophication should concentrate on obtaining measurements of nitrogen rather than phosphorus, as bioassays have demonstrated that the Gilbert Bay is nitrogen limited (Stevens and Gillespie 1976; Wurtsbaugh 1988), as are most saline lakes. Nutrient limitation in the less-saline Farmington Bay, however, likely alternates between nitrogen and phosphorus limitation, depending on annual and decade-long cycles in salinity (Wurtsbaugh and Marcarelli 2004b). This complicates the interpretation of nutrient loading information and possible management strategies to reduce the problem in Farmington Bay.

References

Austin, L. 1993. Unpublished hydrology data. Assistant Director, Utah DWQ.

Cooke, D.G., E.B. Welch, S.A. Peterson, and P.R. Newroth. 1993. Restoration and Management of Lakes and Reservoirs. 2nd ed. Lewis Publishers, Boca Raton, FL.

Dodds, W.K. 2002. Freshwater Ecology Concepts and Environmental Applications. Academic Press, San Diego.

Dorm, M.D., B. Herut, and R.F.C. Mantoura. 2004. Nutrient budget of the eastern Mediterranean: Implications for phosphorus limitation. Limnol Oceanogr. 49: 1582-1592.

Herut, B., R. Collier, and M.D. Krom. 2002. The role of dust in supplying nitrogen and phosphorus to the South East Mediterranean. Limnol. Oceanogr. 47: 870-878.

Loving, B.L., K.M. Waddell, and C.W. Miller. Water and Salt Balance of Great Salt Lake, Utah, and Simulation of Water and Salt Movement through the Causeway, 1987-98. 2000. U.S. Geological Survey. Utah Department of N.R. Division of Forest Fire and Land. Tooele County and Salt Lake City, Utah.

Mason, C.F. 2002. Biology of Freshwater Pollution. 2002. 4th ed. Pearson Education Limited. England.

Redfield, G.W. 2002. Atmospheric deposition of phosphorus to the Everglades: Concepts, constraints, and published deposition rates for ecosystem management. The Scientific World 2: 1843–1873.

Stephens, D.W. and D.M. Gillespie. 1976. Phytoplankton production in the Great Salt Lake, Utah, and a laboratory study of algal response to enrichment. Limnol. Oceanogr. 21: 74-87.

Wurtsbaugh W. A. 1988. Iron, molybdenum and phosphorus limitation of N2 fixation maintains nitrogen deficiency of plankton in the Great Salt Lake drainage (Utah, USA). Verh. Internat. Verein. Limnol. 23: 121-130.

Wurtsbaugh, W.A. 1992. Food-web modification by an invertebrate predator in the Great Salt Lake (USA). Oecologia 89: 168-175.

Wurtsbaugh, W.A., A.M. Marcarelli, D. Gross, J. Moore, C. Christison, S. Kircher, and S. Bates. 2002. Comparative analysis of Pollution in Farmington Bay and the Great Salt Lake, Utah. Aquatic Ecology Laboratory Class Project 2001. College of Natural Resources, Utah State University. 21p.

Wurtsbaugh, W.A. and A. M. Marcarelli. 2004a. Phytoplankton and zooplankton in Farmington Bay and the Great Salt Lake, Utah. Report to the Central Davis Sewer Improvement District, Kaysville, Utah. 57 p.

Wurtsbaugh, W.A. and A. M. Marcarelli. 2004b. Analysis of phytoplankton nutrient limitation in Farmington Bay and the Great Salt Lake. Report to the Central Davis Sewer Improvement District, Kaysville, Utah. 57 p.

Vollenwider, R.A. 1971. Scientific fundamentals of the eutrophication of lakes and flowing water, with particular reference to nitrogen and phosphorus as factors in eutrophication. Paris. Organization for Economic Co-operation and Development.

Figures

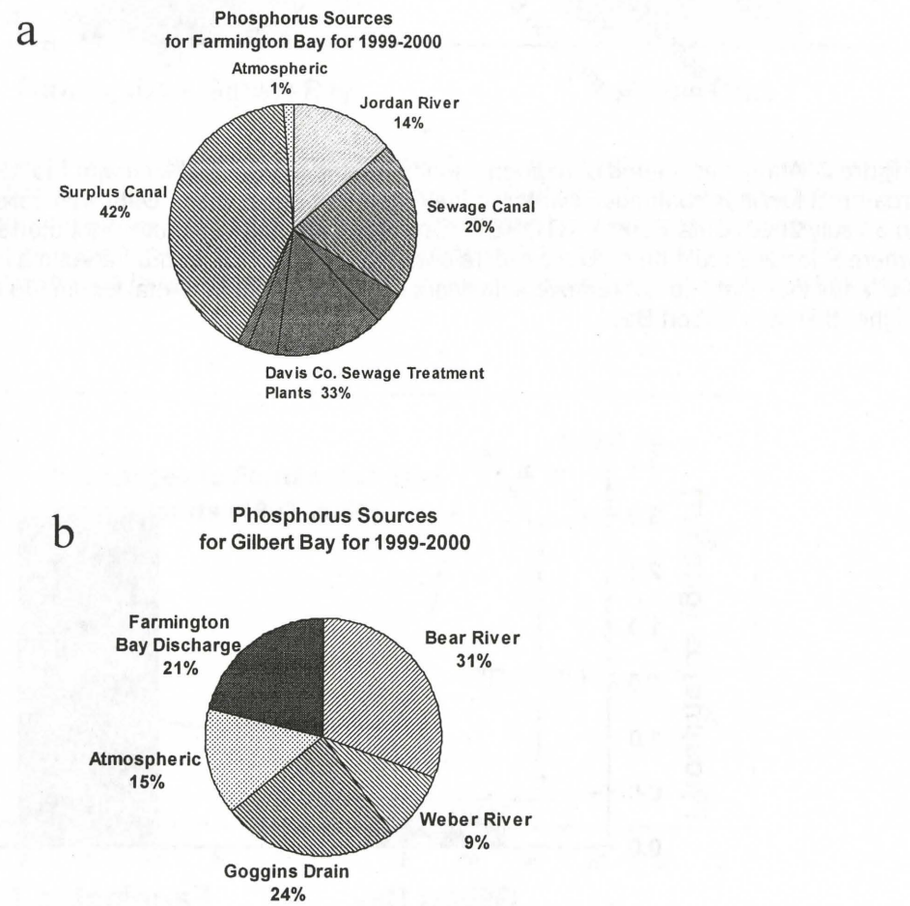

Figure 1. a) Proportions of annual phosphorus for Farmington Bay of the Great Salt Lake, Utah. Davis county sewage includes the South Davis South Wastewater Treatment Plant, the South Davis North Wastewater Treatment Plant, the Central Davis Wastewater Treatment Plant, and the North Davis Wastewater Treatment Pant. b) Proportional phosphorus loading to Gilbert Bay. Data was collected from 1 Aug 1999 to 31 July 2000.

Figure 2. Annual estimated phosphorus loads (metric tons yr^{-1}) of all major tributaries and wastewater treatment facilities contributing water to the Great Salt Lake, Utah. Data was collected from 1 Aug 1999 to 31 July 2000. Data Source: STORET. Down arrows represent known tributaries that flow into wetlands where P loads should be reduced before entering the lake. Other tributaries may flow through small wetlands that likely do not remove significant nutrients. Note that total loading to Farmington Bay is higher than into Gilbert Bay.

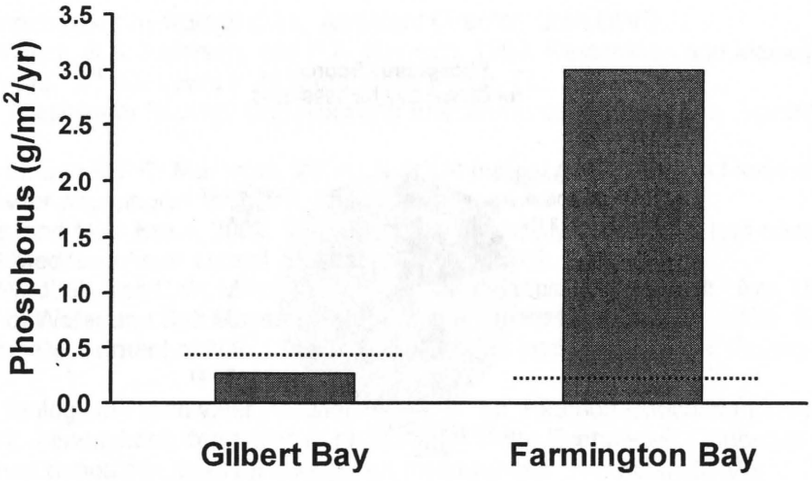

Figure 3. Areal loading rates of phosphorus for Gilbert and Farmington Bays of the Great Salt Lake estimated for 1999-2000. The total loads shown in Figure 2 are diluted across the large Gilbert Bay so that areal loading in Gilbert Bay is far lower than in the smaller Farmington Bay. Dashed lines show the approximate levels estimated to cause severe eutrophication (Cooke et al. 1993).

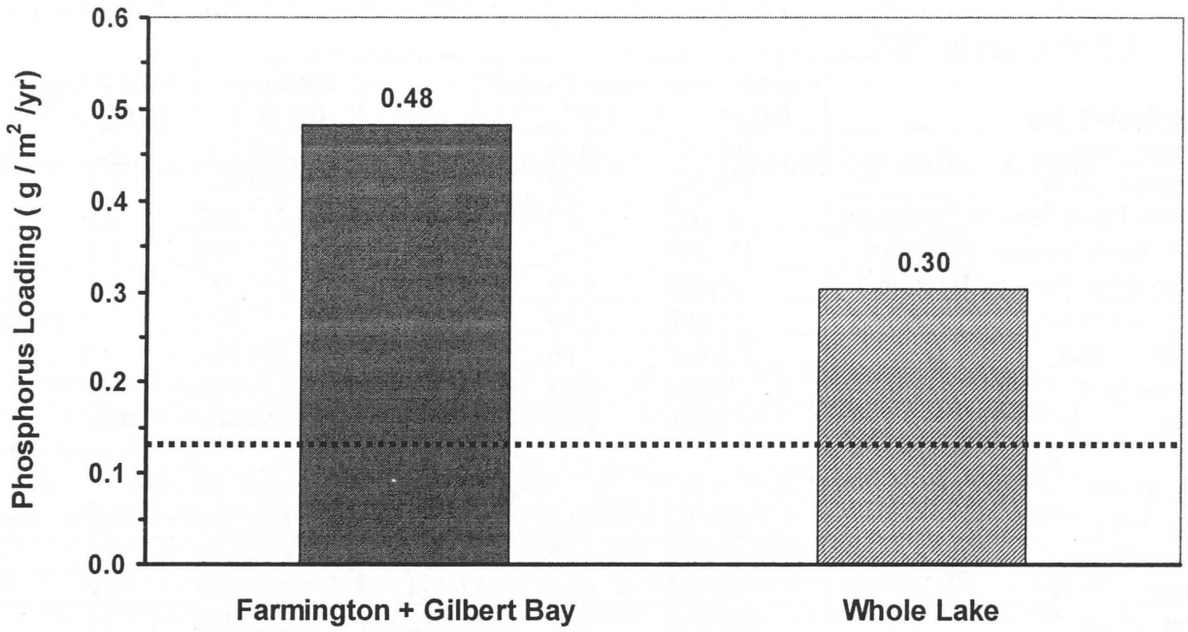

Figure 4. Estimated areal phosphorus loading to the Great Salt Lake if the automobile causeway were removed and nutrient loads were distributed across Farmington + Gilbert Bays (left), and if both the automobile and the railway causeway were removed (right). The dashed line shows the approximate level for dangerous loading leading to eutrophication (Vollenweider 1971). Note that although phosphorus levels are shown here to demonstrate potential issues, the open basin of the Great Salt Lake is nitrogen limited.

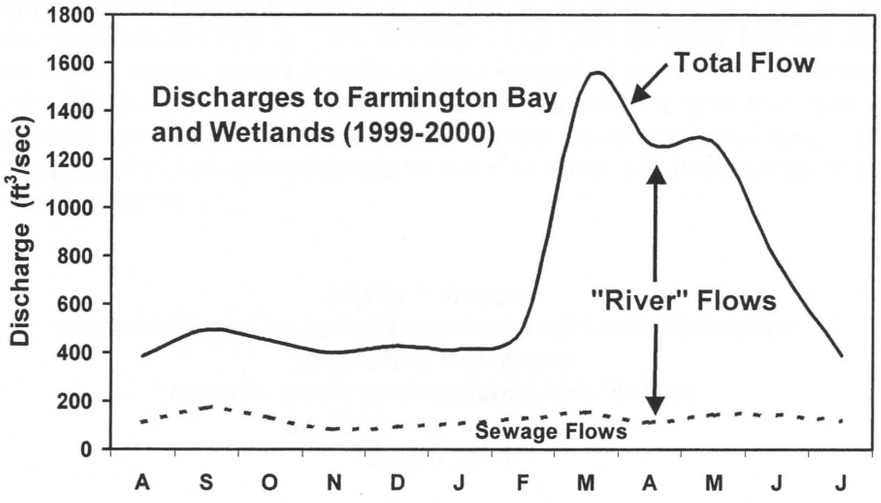

Figure 5. Estimated water inflows from rivers and sewage effluents entering Farmington Bay. All of the river input (Jordan River and Surplus Canal) enters through wetlands and evaporative losses would decrease the actual amount entering the bay. Sewage effluents include those from Davis County treatment plants and the Sewage Canal entering from Salt Lake City.

Tables

Table 1. Estimated phosphorus loading of Farmington and Gilbert Bays of the Great Salt Lake from Aug. 1, 1999 to July 31, 2000.

Farmington Bay	**Phosphorus (kg yr^{-1})**	**Areal P Load (g m^{-2} yr^{-1})**	**%**	**Nitrogen (kg yr^{-1})**	**Areal N Load (g m^{-2} yr^{-1})**	**%**
Jordan River	106,000	0.41	14	1,166,000	4.5	16
Sewage Canal	158,000	0.61	20	1,106,000	4.3	15
Central Davis Sewage Treatment	31,000	0.12	4	217,000	0.8	3
North Davis Sewage Treatment	111,000	0.43	14	777,000	3.0	11
South Davis Sewage Treatment	31,000	0.12	4	217,000	0.8	3
South Davis South S. Treatment	11,000	0.04	1	77,000	0.3	1
Surplus Canal	325,000	1.26	42	3,575,000	13.8	49
Atmospheric	10,000	0.04	1	110,000	0.4	2
Total	**775,000**	**2.99**		**7,245,000**	**28.0**	
Gilbert Bay						
Bear River	190,000	0.08	31	2,090,000	0.9	31
Weber River	58,000	0.02	9	638,000	0.3	9
Goggins Drain	150,000	0.06	24	1,650,000	0.7	24
Atmospheric	92,000	0.04	15	1,012,000	0.4	15
Farmington Bay Discharge	131,000	0.06	21	1,441,000	0.6	21
Total	**622,000**	**0.26**		**6,831,000**	**2.9**	

Table 2. Permissible loading levels for total phosphorus and nitrogen (g m^{-2} yr^{-1}; Vollenweider 1971).

Mean depth up to	**Permissible loading of P**	**Permissible loading of N**	**Dangerous loading in excess of P**	**Dangerous loading in excess of N**
5m	0.07	1.0	0.13	2.0
10m	0.10	1.5	0.20	3.0

Chapter 2

Nitrogen fixation across a salinity gradient: Death by salt... a true story

Summary

Nitrogen Fixation is an important aspect of the nitrogen cycle in saline lakes where nitrogen is often a growth-limiting nutrient. In the Great Salt Lake the primary nitrogen-fixing cyanobacteria is believed to be *Nodularia* sp. but some recent work has shown that it may not fix nitrogen in salinities above 7%. To investigate how nitrogen fixation and algal populations changed with salinity, we examined a 20-km long "gradient" from the less-saline Farmington Bay (3.5%) into Gilbert Bay (16%) in the Great Salt Lake in late September 2004. We were unable to sample fine scale changes in salinity as the gradient changed sharply from 3.5% to 15% at the transition from Farmington Bay to Gilbert Bay. Chlorophyll levels were 125 μg L^{-1} in Farmington Bay and declined rapidly as the plume of water from the bay was diluted into the larger lake were concentrations were only 1 μg L^{-1}. Nitrogen fixation rates measured with acetylene reduction indicated that nitrogen fixation was high (2 μg L^{-1} h^{-1}) in Farmington Bay but declined rapidly to zero in Gilbert Bay. Stable isotope analyses of ^{15}N in seston showed that levels were $\delta^{15}N$ +5 in Farmington Bay, but increased to $\delta^{15}N$ +7-9 in Gilbert Bay, supporting the observation that nitrogen fixation may contribute a significant amount of nitrogen to plankton in Farmington Bay. To test the importance of salinity for controlling nitrogen fixation, we set up a laboratory experiment to measure changes in nitrogen fixation over time when *Nodularia* were exposed to various salinity levels, including ones thought to be above their tolerance range. Laboratory bioassays showed that at 3.5% N-fixation was fairly constant but that *in vitro* nitrogen fixation ended in less than 4 h when salinity increased above 10%. Further study will be needed to identify whether these same conditions occur when a large and even gradient of salinity exist during a more extensive plume event. Stable isotope analysis results were promising and require study on a larger scale to determine the true importance of nitrogen fixation to this saline system.

Justin Robinson
Department of Aquatic Watershed and Earth Resources
Utah State University

Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 Dec 2004

Introduction

Nitrogen has been identified as the limiting nutrient for phytoplankton population growth in many water bodies, both fresh and marine (Marcarelli et al. 2003, Wurtsbaugh 1988, Wurtsbaugh and Marcarelli 2004, Howarth et al.1988, Dodds 2002, Vincent et al. 1984). If nitrogen is limiting in the Great Salt Lake, nitrogen-fixing cyanobacteria can play a vital role in the assimilation of atmospheric N_2 gas into organically available nitrogen compounds (NH_4), which can subsequently be redistributed throughout the food web, increasing total productivity (Wurtsbaugh and Marcarelli 2004). Increasing biologically available nitrogen might increase phytoplankton production, an important food resource for brine shrimp, *Artemia franciscana*, a very important economical and ecological resource of the Great Salt Lake (Nicholson and Marcarelli 2004).

A nitrogen fixing cyanobacteria found in Great Salt Lake, *Nodularia* sp., has been shown to tolerate a limited range of salinity (Fernandes et al. 1993, Marcarelli et al. 2003, Wurtsbaugh and Marcarelli 2004). *Nodularia's* ability to fix N has been demonstrated to decline as salinity increases (Carter 1971). At salinity levels of about 7% (all salinity measurements in this report are weight by volume) nitrogen fixation ends and population densities only increase at salinities below 3-6% (Marcarelli et al. 2003, Wurtsbaugh and Marcarelli 2004). The south arm of the Great Salt Lake (Gilbert Bay) typically has salinity levels around 17%. Farmington Bay is mostly isolated from Gilbert Bay by a road causeway and Antelope island, yet it receives large inflows of freshwater from the Jordan River and sewage canals. The causeway has been breached to allow water to exit into Gilbert Bay. Because of this isolation and "fresh" water inputs, Farmington Bay has salt concentrations much lower than Gilbert Bay. Typical salinity in Farmington Bay ranges from 2-10% depending on the amount of inflow, water exchange with Gilbert Bay, and the rate of evaporation.

As Farmington Bay water enters into Gilbert Bay, high nutrient levels and lower salinity from Farmington Bay cause algae blooms that can extend many kilometers into Gilbert Bay (Figure 1). As the plume extends into Gilbert Bay, the fresh and saline waters mix and salinity increases. Since nitrogen is important in the Great Salt Lake and the N-fixing cyanobacteria only appear to perform N-fixation at lower salinities, it is important for managers to understand what is happening in the mixing waters between Farmington and Gilbert Bays. Of particular interest would be the functionality and N-fixing ability of *Nodularia* sp. along the salinity gradient from the breach of the causeway to the high salinity of undiluted Gilbert Bay water.

Three experiments were designed to identify N-fixation rates in the field and in the laboratory as wells as identify sources of nitrogen in both bays using nitrogen isotopes. These experiments will help understand the rate at which N_2 gas is being assimilated from the atmosphere. Our isotope data will help us identify nitrogen isotopes because the ^{15}N signatures should be high in samples that have been using organic nitrogen when compared to those that are fixing atmospheric nitrogen. Ratios of ^{15}N:^{14}N are higher in organic nitrogen bearing compounds than in N_2 gas, because N_2 gas has been established as the standard with ^{15}N:^{14}N isotopes in atmospheric gas set to zero (Peterson and Fry 1987).

We predicted that N-fixation would decrease as salinity increases, that in low salinity Farmington Bay stable isotope analysis would show that there would be more atmospheric nitrogen assimilated in phytoplankton than in Gilbert Bay's high salinity waters, and that nitrogen fixation and plankton abundance would be higher at field sites with low salinity.

Study area and Methods

We established a transect starting at the at the causeway breach and extending into Gilbert Bay for 20 km, and into Farmington Bay approximately 1 km (Figure 2). It encompassed all available salinity ranges of the two bays, including one site in unstratified Gilbert Bay and

another at a site in Farmington Bay that was stratified by a salt wedge. The established transect was broken into 11 evenly spaced segments of 1.8 km. At each site limnological parameters were gathered including Secchi depth, salinity, temperature. Water samples were collected from just below the surface (0.2 m) with a Van Dorn bottle and transported to the lab for further analysis. Locations and characteristics of each site are shown in Table 1.

Chlorophyll *a* was measured as a metric for primary production. We filtered 50 mL of the sample water through 25-mm Millipore AP40 glass filters with a pore size of 1 µm under low vacuum and placed in a freezer for one month prior to analysis. Additional samples for chlorophyll analysis were taken at both ends of the transect by the AWER 4510 class and analyzed within one week of collection. The filtered samples were extracted in 95% ethanol and processed with a fluorometer (Welschmeyer 1994). Chlorophyll analysis of the field transect samples yielded unrealistically low concentrations, probably as the result of the prolonged storage of salt-saturated filters that did not freeze. Consequently, we compared concentrations from the transect with those analyzed by the class within one week to calculate a correction factor. This factor was then applied to all of the values from the transect and the resulting values are expressed as "Chlorophyll Index Values".

N-fixation was determined using acetylene reduction assays (Capone 1993). Nitrogen fixation assays for the transect study were conducted in the laboratory 12 h after field samples were collected. They were incubated at 20ºC and at a light intensity of 120 µE m^{-2} s^{-1} for 2 hours. Standards were prepared and incubated with the samples. Acetylene reduction rates were converted to nitrogen fixation rates using an assumed 3:1 ethylene: nitrogen molar conversion ratio (Capone 1993). Statistical analysis consisted of a two-way analysis of variance (ANOVA) of salinity versus N-fixation using SAS Version 8e; significance was evaluated at $p<0.05$.

To examine the effects of salinity on nitrogen fixation by *Nodularia*, cells were exposed to salinities ranging from 3.5 to 17% in the laboratory and fixation was assayed four times over 24 hours. This experiment was designed to mimic conditions that could occur in the lake with rapid mixing at the interface between the two bays, such as during severe storm events. Farmington Bay water containing *Nodularia* sp. was mixed with concentrated saline solutions mimicking the component salts of the Great Salt Lake. NaCl and $MgSO_4$ were dissolved in deionized water in a ratio of 8.7:1.3 respectively, imitating the major salt components of the Great Salt Lake. The water collected from Farmington Bay was then mixed with the proper amount of salt enriched water to yield three mixtures with salinities of 5.8%, 10% and 17%. The 5.8% salinity was selected because it is just below what has been identified as a threshold value for *Nodularia* nitrogen fixation (Marcarelli et al. 2003, Wurtsbaugh and Marcarelli 2004). The 17% salinity imitated Gilbert Bay water during field sampling, and 10% was chosen as a midpoint. Undiluted Farmington Bay water with a salinity of 3.5% was included as a control. The water samples from Farmington Bay were held for 14 days in the laboratory at 20ºC and 200 µE m^{-2} s^{-1} prior to the experiment. N-fixation was measured with acetylene reduction assays as described above at intervals of 0, 1, 2, 4 and 24 hours to determine salinity effects on N-fixation over time. Statistical analysis consisted of a two-way ANOVA of salinity versus N-fixation as described above.

Samples for stable isotope analysis were collected in Farmington Bay and at 1.6 and 19.6 km from the causeway breach to determine ratios of ^{15}N:^{14}N over the salinity gradient. These points included the extreme ends of the transect, and the point (1.8 km) we estimated as the most heterogeneous water of the two bays. Water samples collected above were filtered through 25-mm Millipore AP40 glass fiber filters until they clogged (70 mL in Farmington Bay, 515-532 mL in Gilbert Bay). The filters and seston were dried at 60°C for 24h, encapsulated, and shipped to U.C. Davis for N and C isotope analysis in a mass spectrometer (Peterson and Fry 1987). Differences in delta values and nitrogen concentration between sites were analyzed using a one-way ANOVA.

Results

Analysis of the algal plume–Chlorophyll *a* analysis indicated high algal production variability along the transect (Figure 3). In the hypereutrophic Farmington Bay, phytoplankton concentrations were so high (120 µg L^{-1}) that light was likely limiting. However, chlorophyll levels were nearly 5 times higher in the deep strata of Farmington Bay than in the surface waters, probably as the result of sedimented phytoplankton accumulating in the deep brine layer. Chlorophyll concentrations decreased markedly along the transect, finally reaching concentrations near 2 µg L^{-1} 3-5 km from the breach. Variability in replicate samples was high, probably as a result of errors induced by prolonged storage. Secchi depth, which is linked to productivity (Carlson 1977), increased from 0.39 m in Farmington Bay, to 1-2 m in the plume, to 3.1 m at the end of the sampling transect in Gilbert Bay (Figure 4). The variation in Secchi depth in Gilbert Bay near the causeway breach is associated with phytoplankton and dissolved organic mater from Farmington Bay water mixing with low production Gilbert Bay water; this variation is labeled as "Plume" in Figure 4.

Sampling at field sites demonstrated a strong connection between salinity and nitrogen fixation (Figure 5). Samples collected in salinities lower than 4% in Farmington Bay fixed nitrogen at very high rates while all other samples in Gilbert Bay had no discernable rates of fixation.

Isotopic nitrogen signatures in Farmington Bay were lower than in Gilbert Bay (Figure 6). Delta ^{15}N ratios in Farmington Bay were near 5, and they increased to 7.6-9 in Gilbert Bay. The ratios at the three sites were significantly different (ANOVA; $F_{1,4} = 28.22$, $p = 0.006$). Particulate nitrogen and carbon concentrations determined by mass spectrometry were 6-7 times higher in Farmington Bay than in Gilbert Bay (Figure 7).

In vitro *analysis of salinity effects on nitrogen fixation*–N-fixation by the Farmington Bay plankton community decreased rapidly, and even ceased, with increased salinity. Results show that control samples fixed N at rates significantly higher than samples with increased salinities ($F= 6.91$, $P=0.003$; Figure 8). When salinity was increased to 5.8% fixation was still present but proceeded at a depressed rate. In higher salinities (10 and 17%) N-fixation decreased to near zero within 2 hours of the salinity increase. Upon initial exposure to salinities of 5.8% the plankton from Farmington Bay stopped fixing N but resumed fixation after a short recovery period at a lower rate than the control. The two-way ANOVA showed that N-fixation was significantly greater ($F=5.03$, $P=0.007$) at time 0 and 1 than at any other time, but there was no difference due to interactions between time and salinity ($F= 0.50$, $P=0.87$).

Discussion

The transect analysis of chlorophyll, Secchi depth and particulate N and C demonstrate the marked changes that occurred between Farmington and Gilbert Bay. We found, however, at the time of the study the algal plume from Farmington Bay did not extend far into Gilbert Bay. This was not surprising, given that discharges out of Farmington Bay at the time of the analysis were only about 6 m^3 sec^{-1} (David Naftz, USGS, pers. comm.). Under higher flows, a larger plume might be expected.

Both field and laboratory assays show that N-fixation rates of the Farmington Bay plankton community decrease rapidly with increasing salinity. Further, they stop fixation completely when salinity increases high enough. Implications are that in this nitrogen-limited lake (Wurtsbaugh 1988), when plankton are carried from Farmington Bay into Gilbert Bay they will no longer fix nitrogen. This limitation of nitrogen fixation by salinity levels has been shown in another saline lake and has been hypothesized to perpetuate nitrogen limitation in that system (Herbst 1998). The nitrogen fixing organism in the assays was presumed to be *Nodularia sp.*

(probably *Nodularia spumigena;* Felix and Rushforth 1979), as this was the only heterocystous cyanobacteria encountered in the phytoplankton analyses done by the AWER 4510 class. We did not, however, count large numbers of cells and it is consequently possible that some other nitrogen fixing organisms were present.

Isotopic analyses indicated that nitrogen fixation in Farmington Bay may contribute a significant amount of nitrogen to the system. Delta signatures are the ratio of the heavy isotope ^{15}N to ^{14}N. By definition, the isotopic composition of atmospheric N_2 is delta zero. The longer nitrogen spends in organic compounds, the more opportunity it has to gain a neutron and become heavy ^{15}N (Peterson and Fry 1987). The analysis of our samples showed that Farmington Bay seston had lower delta values than Gilbert Bay; this likely indicates that N-fixation is sufficient to lower the isotopic content of the plankton. The water that enters Farmington Bay is high in nutrients from sewage effluent and urban runoff from Salt Lake and Davis counties. Luecke et al. (2004) found that delta ^{15}N values in seston increased from ~1 to nearly 5 as sampling progressed from pristine headwaters through agricultural and urban use lands. When tertiary treated wastewater was added, delta values increased to over 13 for seston. This suggests that waters entering Farmington Bay should have high delta ^{15}N, because they are rich in waste. Because waters leaving Farmington Bay and entering Gilbert Bay have low deltas, another factor must be decreasing the delta values Farmington Bay. The only possible factor decreasing delta values in Farmington Bay is N-fixation by cyanobacteria. It stands to reason that if delta ^{15}N values of seston in water leaving Farmington Bay are half of what we expect the waters entering Farmington Bay contain, then organisms there are fixing as much N_2 gas as they are using from dissolved sources. In addition to plankton, it is possible that nitrogen fixation is occurring in periphyton of Farmington Bay, but this possibility has not been examined.

Our initial desire to sample across a extended gradient of mixing waters from Farmington Bay and Gilbert Bay was not possible during this study due to low exchange rates and weather-related mixing events. Since we have good field data on the extreme salinities in the two bays and lab bioassays of the gradient, a study mimicking this one should be carried out when it can be established that the large mixing zone or "plume" is present in Gilbert Bay. Further, the stable isotope analysis study should be expanded to span the length of Farmington Bay to attempt to verify the change in delta ^{15}N values between river inflows and its discharge into Gilbert Bay presumably caused by nitrogen fixation.

References

Capone, D.G. 1993. Determination of nitrogenase activity in aquatic samples using the acetylene reduction procedure. Pp 621-631 In P.F. Kemp, B.F. Sherr, E.B. Sherr and J.J. Cole (eds). Handbook of Methods in Microbial Ecology. Lewis Publishers, Boca Raton, FL.

Carlson, R. E. 1977. A trophic state index for lakes. Limnol. Oceanogr. 22: 361-369.

Carter, C. K. 1971. Some ecological considerations of the Farmington Bay estuary and adjacent Great Salt Lake Park. Salt Lake City, University of Utah.

Dodds, W.K. 2002. Freshwater Ecology. Academic Press, San Diego.

Felix, E.A. and S.R. Rushforth. 1979. The algal flora of the Great Salt Lake, Utah, U.S.A. Nova Hedwigia 31: 163-195.

Fernandes, T.A., V. Iyer, and S.K. Apte. 1993. Differential responses of nitrogen-fixing cyanobacteria to salinity and osmotic stresses. Applied and Environmental Microbiology 59: 899-904.

Herbst, D.B. 1998. Potential salinity limitations on nitrogen fixation in sediments from Mono Lake, California. Interational Journal of Salt Lake Research 7: 261-274.

Howarth, R.W., R. Marino, J. Lane, and J. J. Cole. 1988. Nitrogen fixation in freshwater, estuarine, and marine ecosystems. 1. Rates and importance. Limnol. Oceanogr. 33: 669-687.

Luecke, C., Thompson, B. and Mesner, N. 2004. Use of stable isotopes to assess effects of agricultural land use on aquatic systems in small watersheds. Unpublished.

Marcarelli, A.M., W.A. Wurtsbaugh, B. Albrecht, E. Archer, J. Bassett, M. Beckstrand, M. Hadley, J. Kling, O. Lester, P. Mackinnon, and T-h Ting. 2003. Continuing studies of water quality in Farmington Bay and the Great Salt Lake, Utah. Aquatic Ecology Practicum Class Project 2002. College of Natural Resources, Utah State University. 91 p.

Nicholson B. and Marcarelli A. M. 2004. The paradox of a Great Salt Lake. Southwest Hydrology. 3: 24-25.

Peterson, B.J. and B. Fry. 1987. Stable isotopes in ecosystem studies. Ann. Rev. Ecol. Syst. 18: 293-320.

Welschmeyer, N.A. 1994. Fluorometric analysis of chlorophyll *a* in the presence of chlorophyll b and pheopigments. Limnol. Oceanogr. 39: 1985-1992.

Wurtsbaugh, W.A. 1988. Iron, molybdenum and phosphorus limitation of N2 fixation maintains nitrogen deficiency of plankton in Great Salt Lake drainage (Utah, USA). Verh. Int. Ver. Limnol. 23: 121-130.

Wurtsbaugh, W.A. and A.M. Marcarelli. 2004. Analysis of phytoplankton nutrient limitation in Farmington Bay and the Great Salt Lake. Report to the Central Davis Sewer Improvement District. 57pp.

Vincent, W.F., Wurtsbaugh, W.A., Vincent C.L. and Richerson P.J. 1984. Seasonal dynamics of nutrient limitation in a tropical high-altitude lake (Lake Titicaca, Peru-Bolivia): Applications of physiological bioassays. Limnol. Oceanogr. 29(3): 540-552.

Figures

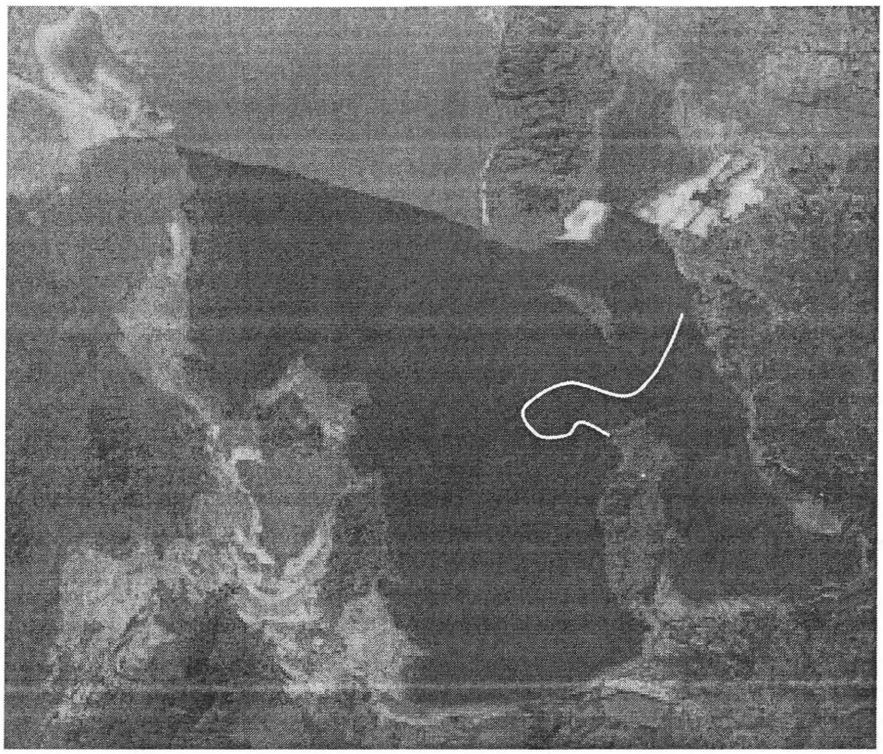

Figure 1. NASA photograph of the Great Salt Lake in September 1992, showing a large plume of algal-rich water extending from Farmington Bay into Gilbert Bay of the Great Salt Lake. Green areas around the shallow margins of the lake may be periphyton rather than phytoplankton. The plume is outlined in white.

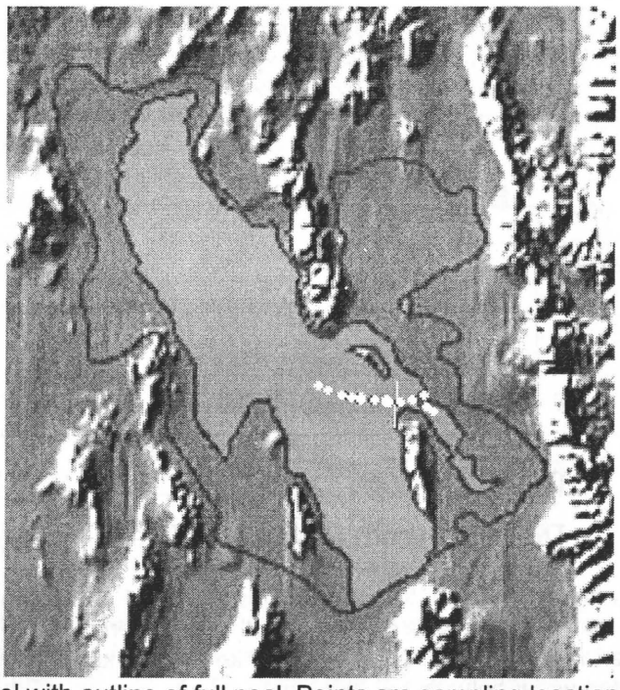

Figure 2. Current lake level with outline of full pool. Points are sampling locations along the transect in Gilbert Bay and in Farmington Bay.

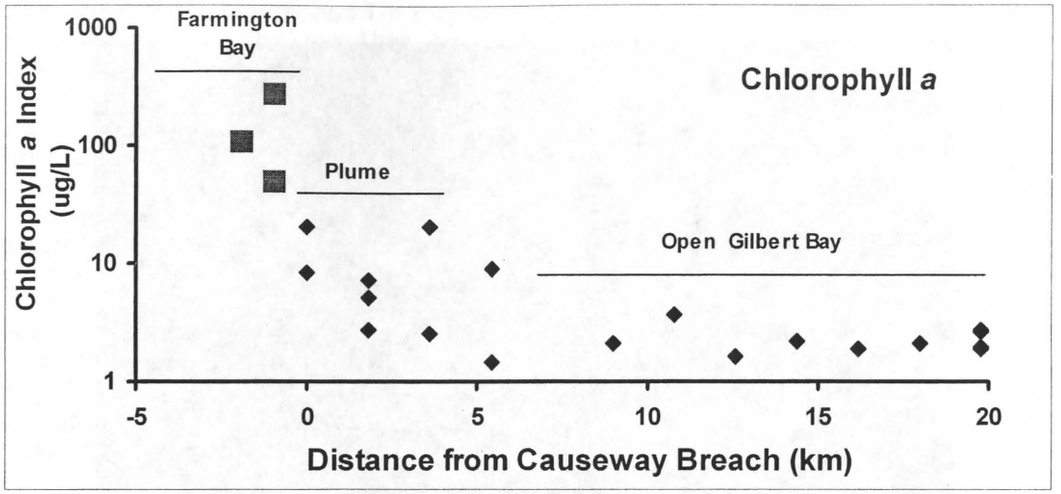

Figure 3. Chlorophyll *a* values from field sites. Distances are scaled to 0 at the Causeway Breach, so that negative values along the transect indicate Farmington Bay sites and positive distances extend into the pelagic zone of Gilbert Bay. An index of chlorophyll was used to compensate for inconsistent results believed to be caused by prolonged storage (see methods). Note the log scale for chlorophyll index values. Sites in Farmington Bay surface waters are shown with square symbols.

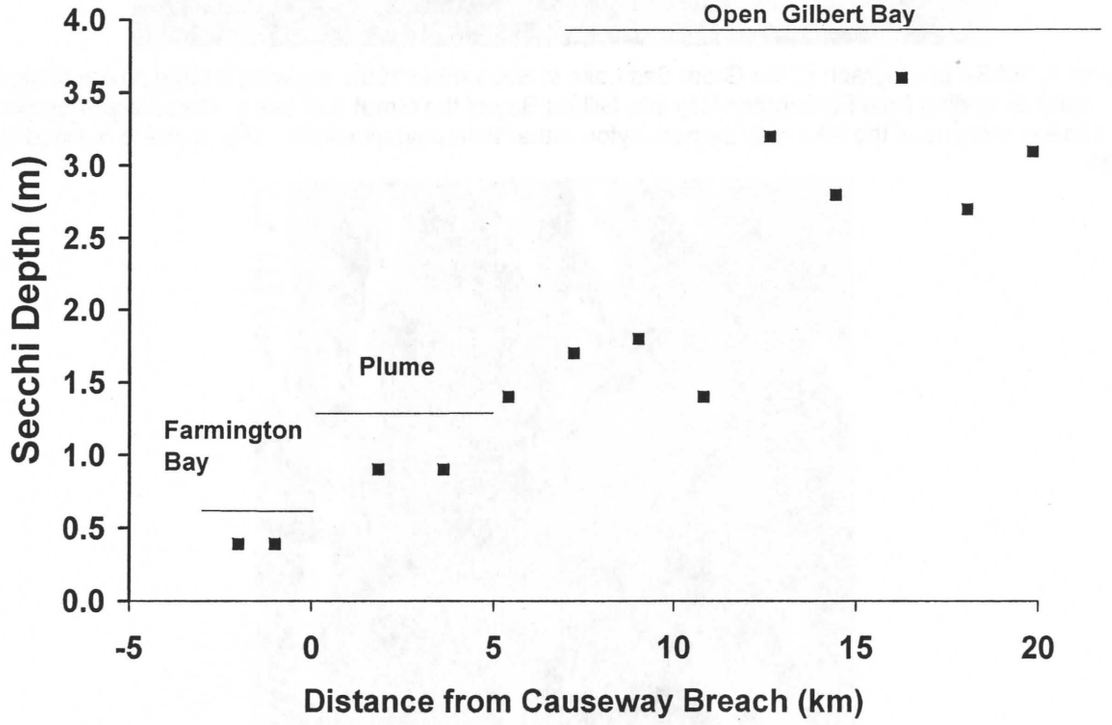

Figure 4. Secchi depth along the transect. Negative values indicate Farmington Bay sites south of the causeway breach. Plume sites indicate where Secchi depth was lowered due to mixed waters of Farmington Bay and Gilbert Bay. Secchi depths from 5.8 km to 10.8 km stations along the transect were greater than the depth of the water column.

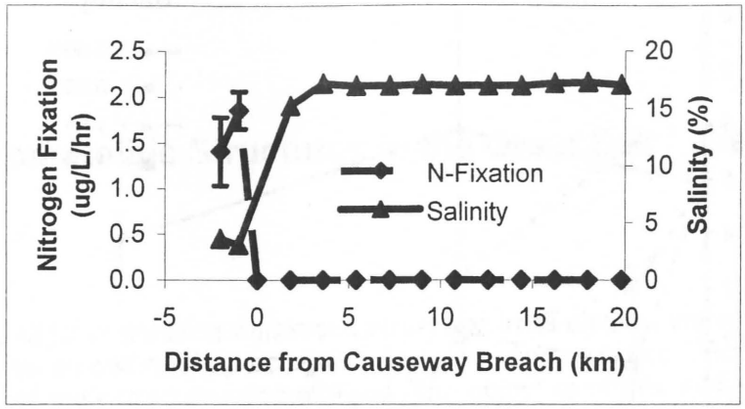

Figure 5. Nitrogen fixation from field samples. Nitrogen fixation (double triangle symbols) levels are high at sites with low salinity (triangle symbols). Left y-axis indicates N-fixation; right y-axis is salinity at the site that samples were collected.

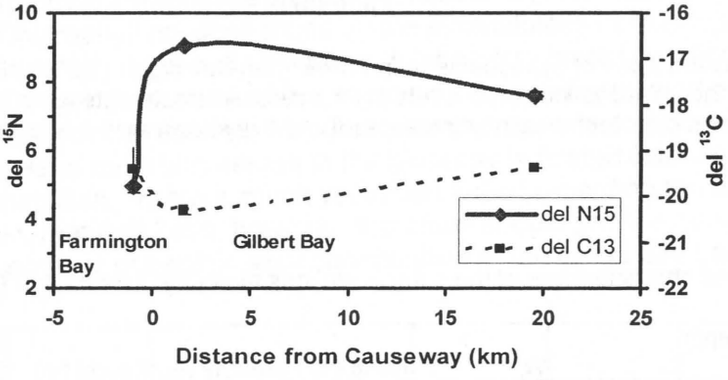

Figure 6. Delta ^{15}N (left axis) and ^{13}C (right axis) signatures from stable isotope analysis of seston in Farmington Bay and Gilbert Bay of the Great Salt Lake (30 September 2004). The X-axis indicates distance from causeway breach (0 km) and the respective bay from which samples were taken. Error bars show ± 1 S.E. when they extend beyond the data points. Farmington Bay samples show significantly lower delta ^{15}N indicating more N-fixation.

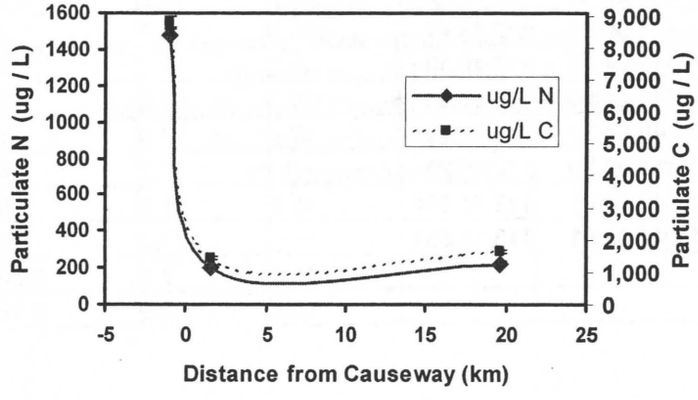

Figure 7. Total nitrogen (left y-axis) and carbon (right y-axis) contained in the seston of Farmington (left x-axis values) and Gilbert Bays (right part of x-axis). Note the scalar differences between the y-axes. Error bars (± 1 SE) are included but may not be visible due to low variances.

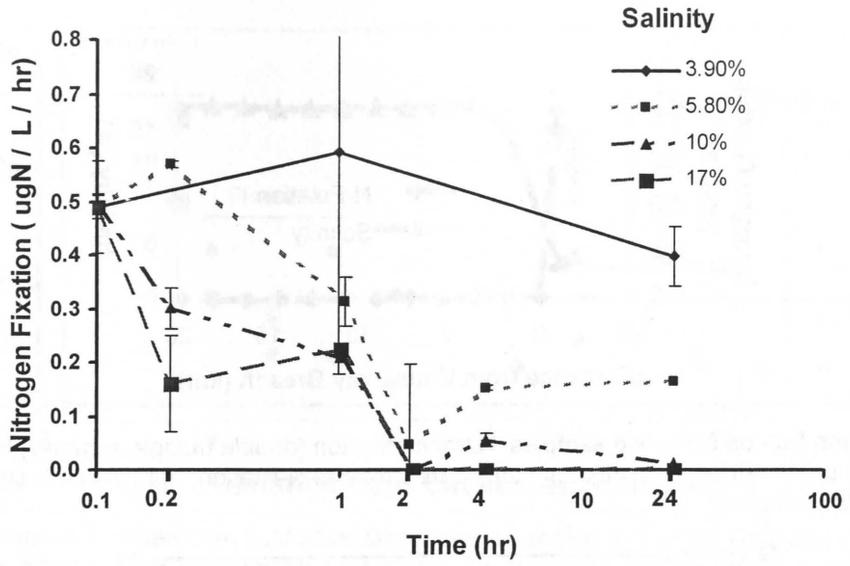

Figure 8. Nitrogen fixation rates of cyanobacteria in Farmington Bay water (3.9% salinity), and in water where salinities were increased to indicated levels at time zero. All treatments were assumed to have the same initial fixation rates prior to the salinity increase. Time 0 is shown as 0.1 hours in this logarithmic plot. Error bars indicate ± 1 S.E.

Tables

Table 1. Locations and characteristics of the sample stations along the Farmington Bay to Gilbert Bay transect.

Station (km)	Lake Depth (m)	N	W	Temp (C)	Salinity (%)	Secchi (m)	Sample Depth (m)
Deep	6.4	41 06.345	112 27.009		17.0		0.25, 6.0
16.2	6.4	41 06.14	112 25.79	18.1	17.1	3.1	0.25
14.4	6.2	41 06.14	112 24.47	18.0	17.3	2.7	0.25
12.6	5.9	41 05.69	112 23.09	18.0	17.2	3.6	0.25
10.8	5.2	41 05.48	112 21.98	18.1	17.0	2.8	0.25
9	3.5	41 05..22	112 20.74	17.9	17.0	3.2	0.25
7.2	1.4	41 05.05	112 19.49	18.1	17.0	1.4	0.25
5.4	1.8	41 04.82	112 18.19	18.1	17.1	1.8	0.25
3.6	1.7	41 04.55	112 16.94	18.1	17.0	1.7	0.25
1.8	1.4	40 04.29	111 15.73	17.9	17.0	1.4	0.25
1.8	1.4	41 04.29	112 15.73		17.2	1.4	0.25
WP # 088 –1.5	1.1	41 03.91	112 14.554	19.0	15.3	0.9	0.25
WP # 089 –1.5	1.1	42 03.91	113 14.554		17.0	0.9	0.2, 0.8
FB #1 -0.5	1.0			15.8	3.1	0.39	0.2
FB #2 -0.5	—			16.8	3.6	0.39	0.2

Chapter 3

Ecology of Stromatolitic Structures in the Great Salt Lake, Utah

Summary

The Great Salt Lake contains numerous structures in its shallow waters that are the result of a union between carbonate precipitates and algae communities. These poorly understood structures are known as stromatolites. The objective of this study was to examine the general shape and size, and algal taxa of the stromatolites that exist in the Great Salt Lake, and to determine how they respond to nutrient enrichment. Mean areas of the stromatolites were 0.4 and 0.9 m in Bridger Bay and at the north tip of Antelope Island, respectively. The community structure of the stromatolite samples consisted almost entirely of the cyanobacteria *Aphanothece* sp. Chlorophyll concentrations on the stromatolites from Bridger Bay averaged 44 $\mu g\ cm^{-2}$, but showed a high degree of variability. A laboratory bioassay using pieces of stromatolites from Bridger Bay indicated that there was no significant increase in chlorophyll concentrations for treatments receiving nitrogen, phosphorus, or a combined N+P treatment. Initial and final acetylene reduction assays in the bioassay indicated that there was no nitrogen fixation in the stromatolites. There is much yet to be studied regarding the ecology of stromatolites in the Great Salt Lake, however, this study should provide some insight into the virtually unstudied ecology of benthic algal communities of the Great Salt Lake.

Jonas N. Parker
Department of Aquatic, Watershed and Earth Resources
Utah State University

Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 December 2004

Introduction

Prokaryotic life (bacteria and archaea) has been in existence for roughly 3.5 billion years. These organisms continue to thrive today living in virtually every environment on the Earth. Some prokaryotes are able to fix atmospheric nitrogen, and in turn contribute nitrogen to organisms that are not able to utilize atmospheric nitrogen. Early in the evolution of prokaryotes (approximately 3 billion years ago) photosynthesis became a function of some bacteria; these bacteria are known today as cyanobacteria (Campbell et al. 1999). Because these cyanobacteria are able to photosynthesize, they produce significant amounts of breathable oxygen.

The Great Salt Lake represents a unique ecosystem. This terminal lake has salinity concentrations several times greater than any ocean. While the Great Salt Lake is an extreme environment, several organisms, including many bacteria and archaea can survive there. Certain bacteria within the Great Salt Lake can form mineral deposits (Pedone and Folk 1996). These mineral deposits in the Great Salt Lake are termed *stromatolites* and have been observed in other warm, highly saline water bodies throughout the world (Paerl et al. 1996).

Stromatolites, the product of mat-forming prokaryotic microbes, can be traced to nearly the beginning of prokaryotic life and are the most abundant fossils of the Archean and Proterozoic eras (Awramik 1984). It is during this time (2.5 bya – 570 mya) that certain prokaryotic cells began diversifying and forming different shapes as well as structures of megascopic size (Hoffman 1985). These structures do not occur everywhere, however. Mendelsohn (1976) notes that only certain mineral deposits are associated with stromatolites; carbonate rock ranging from limestone to dolomite is the most commonly associated with stromatolites. Interestingly, the area surrounding the Great Salt Lake consists primarily of marine deposits of the Cambrian and Precambrian periods. Due in part to the geologic conditions, stromatolites thrive in the Great Salt Lake.

The mat-forming prokaryotic microbes that comprise stromatolites consist primarily of microscopic, filamentous, coccoid, calcareous cyanobacteria (Wray 1977). Ginsburg et al. (1971) explained that these filamentous forms of cyanobacteria are typically held together by mucus which is able to trap sedimentary particles that in turn build upon each other. It should be noted that the overall shape and size of a stromatolite is dependent on environmental conditions and the presence of burrowing and/or grazing metazoans as seen in Lake Clifton, Western Australia (Moore and Burne 1994). Halley (1977) noted that metazoans are not present in most Great Salt Lake stromatolites. Because of relatively controlled growth rates and concurrent decay processes, rates of accretion in stromatolites have been observed to be approximately 0.5 mm yr^{-1} (Krumbein et al. 1977).

Since these slow growing stromatolites are comprised primarily of various cyanobacterial communities, it stands to reason that photosynthesis by these cyanobacteria should produce oxygen. According to Hickman (2004) the cyanobacteria that comprised stromatolites in the Archean and Proterozoic eras generated a large amount of the oxygen in the Earth's current atmosphere.

It is known that the saline lakes where stromatolites typically grow have low total nitrogen/total phosphorus ratios because phosphorus is conserved in saline lakes (Evans et al. 1996). Indeed nitrogen is the limiting nutrient in the Great Salt Lake (Wurtsbaugh 1988). Additionally, rates of nitrogen fixation in saline lakes are typically low or negligible because of intolerance to salinity by nitrogen-fixing cyanobacteria (Herbst 1998). However, nitrogen fixation by cyanobacteria on stromatolites has been widely observed in other less saline marine areas, and nitrogen fixation by Great Salt Lake stromatolites has never been measured.

Since little to no research has been conducted regarding the biological and chemical processes of stromatolites in the Great Salt Lake, it was important to examine them. Literature review revealed only one vague and outdated reference of algal community structure of

stromatolites in the Great Salt Lake. Furthermore, due to the uniqueness of the Great Salt Lake, including extreme salinity, shallow water depth, and proximity to hyper-eutrophic and nutrient rich Farmington Bay, it was important to understand the stromatolite response to various nutrient additions. Lastly, while minimal nitrogen fixation was expected from the stromatolites due to the highly saline conditions of the Great Salt Lake, a nitrogen fixation test on stromatolites in the Great Salt Lake was conducted to determine whether this process was occurring.

Methods

Study area–The Great Salt Lake is the largest remnant lake of historic Lake Bonneville, which covered most of Utah in the late Pleistocene. The Great Salt Lake is very shallow with a mean depth of approximately 6.5 meters, depending on annual precipitation. Secchi depths in the Great Salt Lake usually extend just past three meters. Salinity in the Great Salt Lake averages about 13%, but was 16% at the time of my study

Within the Great Salt Lake, there are several islands; the largest island is Antelope Island in the southeastern corner of the lake. In recent years, the southern tip of Antelope Island has become joined to the mainland due to low water conditions. Additionally, a vehicle causeway to Antelope Island State Park now connects the northern tip of the island to the mainland. The vehicular causeway and Antelope Island form a body of water that is almost entirely disconnected from the Great Salt Lake, called Farmington Bay. Farmington Bay is a hyper-eutrophic, shallow water body that receives freshwater from several rivers, sewage canals, and wastewater treatment plants. Secchi depths in Farmington Bay average 0.3 meters. Salinity in Farmington Bay is much less than the Great Salt Lake because of its freshwater inputs. Salinities range between 2 and 9%. Because of a hydraulic head gradient and a small break in the vehicle causeway, nutrient-rich water flows from Farmington Bay into the Great Salt Lake.

According to a modified map by Pedone and Folk (1996; Figure 1) stromatolites in the Great Salt Lake are found along the shorelines of Dolphin, Fremont, Antelope, Carrington, and Stansbury Islands. Using this information, a boat was taken to documented sites at the northern-most point of Antelope Island and a site on the SW shore of Bridger Bay on the western coast of Antelope Island (Figure 1). The Northern Antelope Island site is located at 41° 03'.46 N by 112° 14'.58 W and the Bridger Bay site is located at 41° 02'.36 N by 112° 16'.23 W. Extensive stromatolite communities were found at both sites.

Field sampling–After locating the study sites I took measurements of the stromatolites by walking on top of the stromatolites and on the surrounding sandy substrate. With a meter stick or tape measure I documented the length, width, height, and depth of 40 and 20 stromatolites at the Northern Antelope Island and Bridger Bay sites respectively. For some stromatolites, the shape was described. Stromatolites in both areas appeared to be growing together to form larger units. When taking my measurements I discerned between "individual" stromatolites by referencing a furrow that appeared between the individuals. This sampling methodology was accurate as the water was less than 1-m deep at both locations at the time of measurement. The area of measured stromatolites was estimated by assuming that individuals conformed to circles, ellipses or rectangles.

After taking measurements, I collected approximately 50 stromatolite sub-sections at the Bridger Bay site ranging in size from 20-45 cm^2 for the bioassay. These sub-sections were simply small pieces of the larger parent stromatolite that I was able to break off, largely from the margins of the growth. After breaking off the stromatolite samples, I placed each in 500-ml plastic jars and completely filled each of the jars with water from Bridger Bay. Although the

stromatolites came from slightly different locations, the added water was collected at one location and was filtered through a 153-µm mesh to remove zooplankton.

Laboratory bioassay–The laboratory experiment consisted of and initial sample and 4 treatments: control, nitrogen enriched (N), phosphorus enriched (P), and nitrogen and phosphorus enriched (NP). I randomly chose five replicate stromatolite samples for each of the treatments. For the initial samples, I measured nitrogen fixation on five stromatolites the day after collection as described below, then drained the water and placed them in a freezer to be analyzed for chlorophyll with the final samples. I added nothing to the control samples. For nitrogen treatment, I added NH_4NO_3 to produce a concentration of 1,400 µg N L^{-1}. The phosphorus treatment received $NaHPO_4$ at a concentration of 200 µg P L^{-1}. The nitrogen plus phosphorus treatments received both 1400 µg N L^{-1} and 200 µg P L^{-1}.

The samples in their jars were left on their sides under fluorescent lighting to maximize light exposure. Light intensity measured with a 4π PAR sensor was 120 µE m^{-2} sec^{-1}, approximately equivalent to 6% the intensity of full sunlight. Initially, the lights were left on 24 hours per day. The samples were gently agitated each day and their position under the lights randomized. Air bubbles, which we thought were oxygen, began to form on the stromatolites after two days. Because near saturation of oxygen is not a natural condition, the photoperiod was reduced to 18 light : 6 dark on day four of the bioassay.

After seven days, no visible change was seen in the stromatolites, so another dose of nutrients, equivalent to the first, was added to the samples. After another three days without visual change, the bioassay was ended. First nitrogen-fixation was measured on each sample as described below, then water was drained from each sample and the stromatolite was placed in a freezer to rupture cells and aid in chlorophyll extraction. The next day, 150 ml of 95% ethanol was added to each stromatolite to extract the chlorophyll. The samples were immediately covered and left for approximately 24 hours. After extracting, samples were diluted approximately 100:1 (6.0 ml of ethanol: 0.06 ml extract) and absorbance was measured using a Turner 10 AU fluorometer equipped with a Welschmeyer filter set (Welschmeyer 1994). The absorbance measurements were then normalized for stromatolite surface area using the following formula:

$$\mu gChl.a/cm^2 = \frac{(\mu gChl.a/L) * (liters\ in\ extract) * (dilution\ factor)}{(surface\ area\ of\ sample)}$$

Nitrogen fixation–Nitrogen fixation was estimated using the acetylene reduction assay (Capone 1993). Acetylene gas (C_2H_2) was generated by adding calcium carbide pellets to tap water in flask. To measure the nitrogen fixation rate of the stromatolites, I injected 32-ml of acetylene through a septa into the sample jars containing stromatolite sections (total volume = 500 mL). Samples were then agitated for 1 minute to mix the ethylene with the water covering the sample. The samples were incubated for three hours. After incubation, gas samples were collected in cleaned, re-evacuated Vacutainer® brand vials and stored for later analysis. This process was repeated at the end of the bioassay on all treatment replicates. With each sample run I simultaneously incubated a series of six standards. To each of the six standards I added 50 ml of Gilbert Bay water that was filtered through a 0.8 µm GF/F filter. Known amounts of a 1000 ppm mixture of ethylene and helium were added to the standards and analyzed with the experimental gas samples. The gas samples were later analyzed using an SRI 8610C gas chromatograph equipped with a Poropak-T column and a flame ionization detector. Ethylene content was converted to amount of nitrogen fixed using an assumed 3:1 ethylene to nitrogen molar ratio (Capone 1993).

Community structure and identification–Initial microscope observations of stromatolite samples proved difficult, because of the amount of carbonates in the samples. Subsequently, three different sub-samples of stromatolite were collected and the attached substrate was digested in 1.0 N HCl for approximately 8 h. Following the digestion, most carbonates had been successfully removed. For preservation purposes, 90% of the HCl was decanted from the sample jars and replaced with tap water. 90% of the HCl + tap water solution was then decanted. 50% of the jar was then replaced with tap water and Lugol's solution was added as a preservative. After preservation, I proceeded with observation, identification, and measurements. I counted 500+ individuals per taxa and measured the first ten individuals of each taxa for three different sub-samples. I then followed the same procedure on a sub-sample that had not been digested in HCl, to ensure that no taxa had been digested during the acid treatment.

Results

Size & shape of stromatolites–Mean areas of the stromatolites were 0.4 and 0.9 m in Bridger Bay and at the north tip of Antelope Island, respectively (Table 1; Figure 2). The average height of stromatolites above the surrounding sand was 0.1 m, but I did not estimate how far they extended into the substrate.

Laboratory bioassay–At the end of the 10-day bioassay there were no visible differences among stromatolites in the different nutrient treatments, and these observations are reflected in the measured chlorophyll levels Figure 3). The mean values of chlorophyll *a* concentration seem to show that there was a trend between treatments, with those receiving nitrogen, or nitrogen plus phosphorus having the highest mean chlorophyll levels. Likewise, the initial samples had a mean chlorophyll *a* concentration that is nearly the lowest when compared to other treatments. Note, however, the large standard error bars, indicating very high variance between replicates. A single-factor analysis of variance (ANOVA) demonstrated that there was no significant effect of the nutrient additions on chlorophyll levels ($p = 0.63$; Table 2).

Nitrogen fixation–As explained above, nitrogen fixation rates were expected to be low or negligible, because of the hypersaline conditions of the Great Salt Lake. Nonetheless, the acetylene reduction samples were analyzed (Figure 4). Initial samples appeared to be significantly greater than following treatments, but in actuality the nitrogen fixation rates were all extremely low and the differences between the initial value and those at measured at the end of the bioassay can likely be attributed to laboratory error. Initial, control, and other treatments have virtually zero nitrogen fixation rates.

Community structure and identification–After stromatolite sections were digested with acid, the material that remained was a solid, yet flexible plate of algae embedded in mucilage. I identified only two species of algae from the stromatolites. The first was the cyanobacterium *Aphanothece* sp. At 400 power the cells were tinged green or brown depending on the concentration of Lugol's in the sample solution. The cells embedded in the mucilage were singular and colorless inside the cell wall. The cell contained no visible organelles. Interestingly, the cells were oriented with respect to each other on the microscope slide, which is indicative of a colonial mucus assemblage. All cells were very similar in shape (circular) and size (Table 3), with a mean diameter of 1.4 µm.

One species of green algae was observed on three occasions between six sub-samples, but only once while counting and measuring. The green alga was much larger than the *Aphanothece* sp. and had a bold, green cell wall which is indicative of green algae. The single cells that I observed each contained a large dark red or brown solid object within the cell walls.

Lastly, the cells were not positioned in any order or relation to one another. Subsequently, I can confidently say that 99% of the algae community that comprises stromatolites in the Great Salt Lake is *Aphanothece* sp. with less than 1% being comprised of a species of green algae (Figure 6).

Discussion

My bioassay indicated that the periphyton on stromatolites did not respond to nutrient additions. This is a surprising result, since very few water bodies are not nutrient limited. It should be noted that the proximity of Farmington Bay and its outflow into the Great Salt Lake to our collection sites may be a factor of significance. The waters of Farmington Bay are rich in nitrogen and phosphorus. The outflow of nutrient rich Farmington Bay waters into the Great Salt Lake produces a visible plume that can be seen in aerial photography extending for miles and typically covers the northern coast of Antelope Island. It is quite possible that the sampling site in Bridger Bay, and more likely, the sampling site at Northern Antelope Island could have been inundated with nutrients from Farmington Bay. This surplus of nutrients would subsequently void any bioassay as there is potential for not having a nutrient limited community. A future experiment should be conducted to identify whether water exchange with Farmington Bay plays a role in chlorophyll *a* production given different nutrient additions. It is also possible, given the relatively high nutrient levels in the Great Salt Lake that we did not add enough nutrients or allow sufficient time for the periphyton community to respond to our nutrient additions.

Another previously unmentioned factor that may influence our bioassay results is the presence of algae growing on the underside of the stromatolite. The mean chlorophyll *a* concentrations on the stromatolites was 44 $\mu g\ cm^{-2} \pm 34$ (s.d.). The high variability was possibly due to the fact that some stromatolites had dark material on the underside of the section. Stromatolites in both sampling sites had a fringe along the outer edge where the physical structure was somewhat mushroom-shaped. This shaping can be caused by currents of the lake eroding the older base of the stromatolite. The underside of the overhanging fringe was typically very dark. It is unlikely that this color could be attributed to a microscopic alga assemblage as the underside of the fringe receives little to no sunlight and it seems unlikely that periphyton could grow when completely shaded by the carbonate and algal structures above. Nonetheless, a future study should consider this as potentially influencing a nutrient addition bioassay.

Virtually zero nitrogen fixation was measured in all bioassay treatments. Such results are not surprising since most nitrogen-fixing cyanobacteria have low tolerances to higher salinities (Herbst 1998). Additionally, the dominant species of cyanobacteria on the stromatolite is one that does not fix nitrogen. *Aphanothece* sp. does not contain specialized heterocysts that allow many other cyanobacteria to fix atmospheric nitrogen. While it appears that stromatolites were not fixing nitrogen at our collection sites, it may be interesting to examine the effects of salinity and/or temperature on nitrogen fixation as other stromatolite communities have been known to fix nitrogen under certain conditions (Mehta et al. 2003).

In my study, two sampling sites were chosen that were in relative close proximity to one another. Salinity, depth, substrate, and aspect to the fetch of the Great Salt Lake were similar between the two sampling sites. A much more comprehensive survey needs to be conducted to examine whether or not stromatolite type varies with respect to location. Hoffman (1976) notes that in the vicinity of Shark Bay, Australia (a shallow, hyper-saline, marine, lagoon) there are at least five different types of stromatolites that are formed as functions of tidal current, distance from shore, and water depth. Even though my two sampling sites were in close proximity to each other, I still noticed a slight difference between stromatolites at the two sites. At Northern Antelope Island, stromatolites were more dome shaped and their outer texture was soft. At the Bridger Bay site, stromatolites were more crust-like and had a significantly firmer texture.

Additionally, stromatolites there did not appear to form any uniform shapes. Such differences in hardness may play a role in brine fly (*Ephydra cinerea*) development. As Collins (1980) notes in his studies, *E. cinerea* larvae attach to hard substrates such as stromatolites where they develop up to 88% more successfully than on sand substrates, even though the hard substrates cover only 18% of the lake bottom. Such differences in stromatolite hardness may also be indicative of a difference in algal community structure.

The stromatolites in the Great Salt Lake are likely an important source of production and provide structure for other organisms such as brine flies. There is much yet to be studied regarding the ecology of stromatolites in the Great Salt Lake. My work addressed some ecological characteristics of the stromatolites only on the northern end of Antelope Island, and only in October. This study should provide some insight into the virtually unstudied ecology of benthic communities of the Great Salt Lake, but more work is needed in other sections of the lake and in other seasons.

References

Awramik, S.M. 1984. Ancient stromatolites and microbial mats. Microbial Mats: Stromatolites. Y. Cohen [ed.], MBL Lectures in Biology, Vol. 3. Alan R. Liss, Inc., New York.

Campbell, N.A., J.B. Reece, and L.G. Mitchell. 1999. Biology. Addison Wesley Longman, Inc.

Capone, D.G. 1993. Determination of nitrogenase activity in aquatic samples using the acetylene reduction protocol. Handbook of Methods in Microbial Ecology. P. F. Kemp, B.F. Sherr, E.B. Sherr and J.J. Cole [eds.]. Lewis Publishers, Boca Raton, FL.

Collins, N. 1980. Population ecology of *Ephydra cinerea jones* (Diptera: Ephydridae), the only benthic metazoan of the Great Salt Lake, U.S.A. Hydrobiologia 68: 99-112.

Evans, M.S., M.T. Arts, and R.D. Robarts. 1996. Algal productivity, algal biomass, and zooplankton biomass in a phosphorus-rich saline lake: Deviations from regression model predictions. Canadian Journal of Fisheries & Aquatic Sciences 53: 1048-1060.

Ginsburg, R., R. Rezak and J.L. Wray. 1971. Geology of Calcareous Algae, notes for a short course. The Comparative Sedimentology Laboratory. Division of Marine Geology and Geophysics. University of Miami.

Halley, R.B. 1977. Ooid fabric and fracture in the Great Salt Lake. Sedimentary Petrology 47: 1099-1120.

Herbst, D. B. 1988. Potential salinity limitations on nitrogen fixation in sediments from Mono Lake, California. Hydrobiologia 7: 261-274.

Hickman, M. 2004. Blue-green algae. The Canadian Encyclopedia. Historica Foundation, Toronto.

Hoffman, H.J. 1985. Precambrian carbonaceous megafossils. Paleoalgology, Contemporary Research and Applications. D. F. Toomey [ed.]. Springer-Verlag, New York.

Hoffman, P. 1976. Stromatolite morphogenesis in Shark Bay, Western Australia.Stromatolites. M. R. Walter [ed.]. Developments in Sedimentology, Vol. 20. Elsevier Scientific Publishing Company, New York.

Krumbein, W.E., Y. Cohen and M. Shilo. 1977. Solar Lake (Sinai). 4. Stromatolitic cyanobacterial mats. Limnology and Oceanography 22: 635-656.

Mehta, M.P., D.A. Butterfield and J.A. Baross. 2003. Phylogenetic diversity of nitrogenase (*nifH*) genes in deep-sea and hydrothermal vent environments of the Juan de Fuca Ridge. Applied and Environmental Microbiology 69: 960-970.

Mendelsohn, F. 1976. Mineral deposits associated with stromatolites. Stromatolites. M. R. Walter [ed.]. Developments in Sedimentology, Vol. 20. Elsevier Scientific Publishing Company, New York.

Moore, L. S. and R. V. Burne. 1994. The Modern Thrombolites of Lake Clifton, Western Australia. Phanerozoic Stromatolites II. J. Bertrand-Sarfati [ed.]. Kluwer Academic Publishers, Boston.

Paerl, H.W., M. Fitzpatrick and B.M. Bebout. 1996. Seasonal nitrogen fixation dynamics in a marine microbial mat: Potential roles of cyanobacteria and microheterotrophs. Limnology and Oceanography 41: 419-427.

Pedone, V.A. and R. Folk. 1996. Formation of aragonite cement by nannobacteria in the Great Salt Lake, Utah." Geology 24: 763-765.

Welschmeyer, N. A. 1994. Fluorometric analysis of chlorophyll *a* in the presence of chlorophyll *b* and pheopigments. Limnology and Oceanography 39: 1985-1992.

Wray, J. L. 1977. Calcareous algae. Developments in Paleontology and Stratigraphy, Vol. 4. Elsevier Scientific Publishing Company, New York.

Wurtsbaugh, W.A. 1988. Iron, molybdenum and phosphorus limitation of N_2 fixation maintains nitrogen deficiency of plankton in the Great Salt Lake drainage (Utah, USA). Verh. Int. Ver. Limnol. 23: 121-130.

Figures

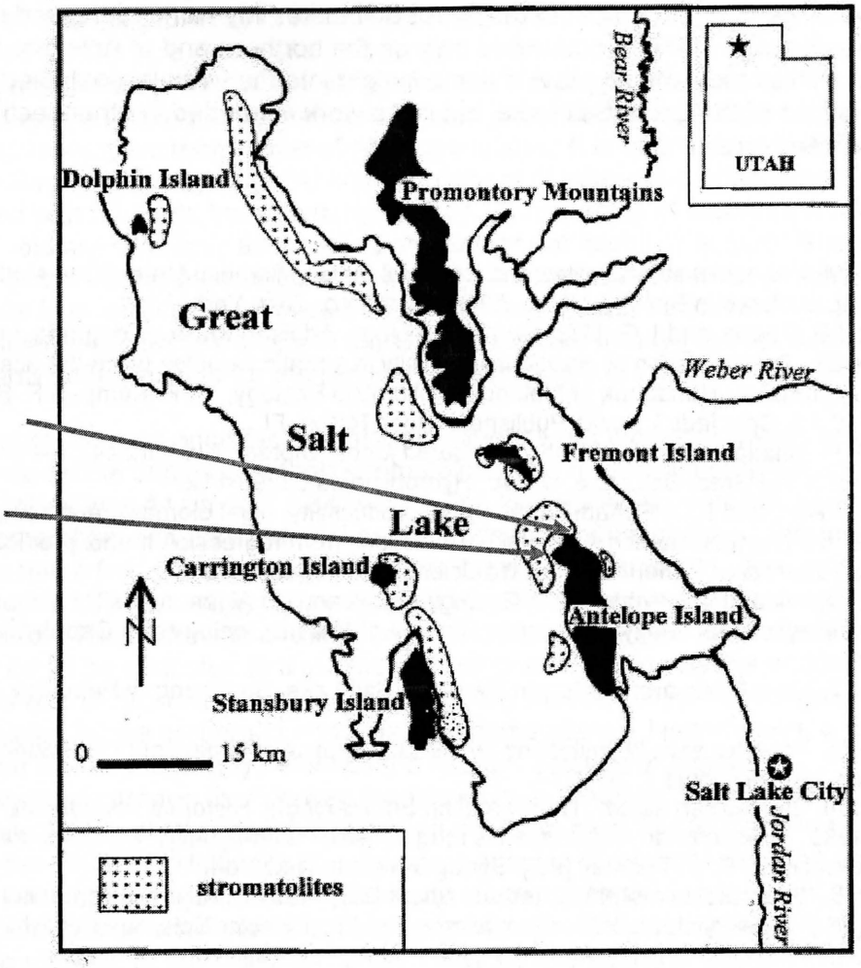

Figure 1. Distribution of stromatolites in the Great Salt Lake and locations of the sampling sites for this study. Adapted from Pedone and Folk, 1996.

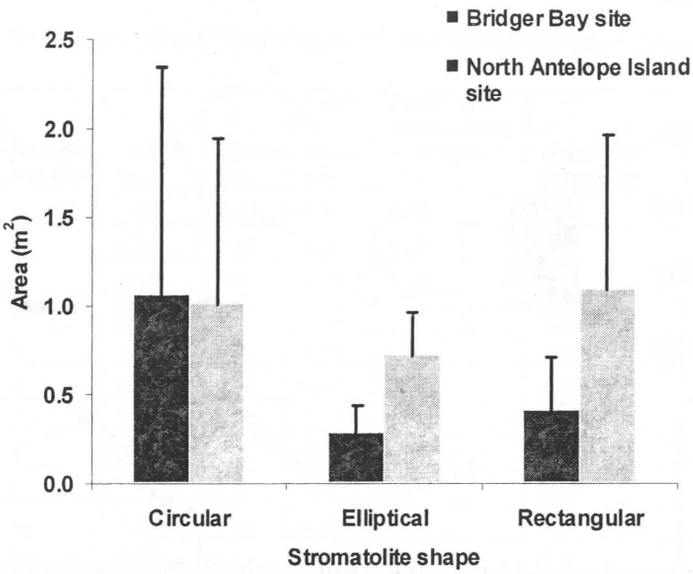

Figure 2. The bars in this graph represent the comparative areas of stromatolites per shape and per location. Error bars represent +1 standard deviation. There are no trends here, but stromatolite areas are typically small. Length and width measurements (taken to determine area) relied on furrows that appeared between individual stromatolites as references.

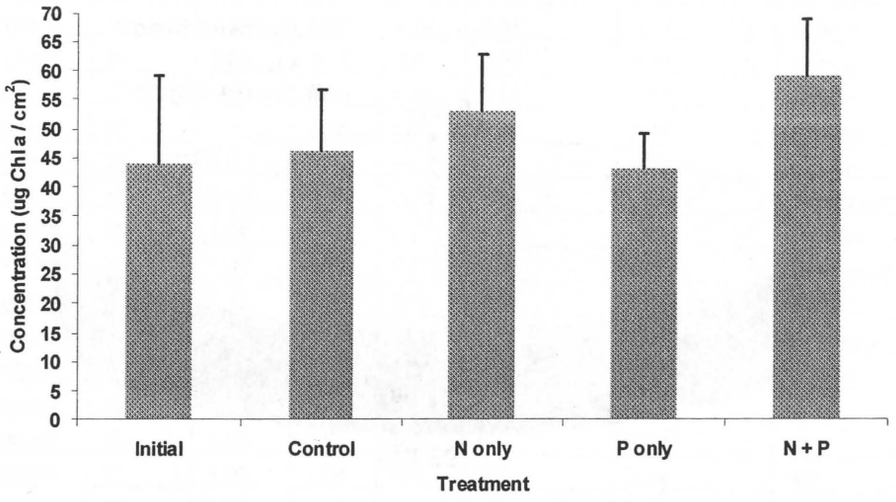

Figure 3. Bars show the mean chlorophyll α concentration averaged for the five replicate samples per treatment in the bioassay experiment. Note that chlorophyll α concentrations have been normalized for stromatolite surface area. Error bars represent +1 standard error.

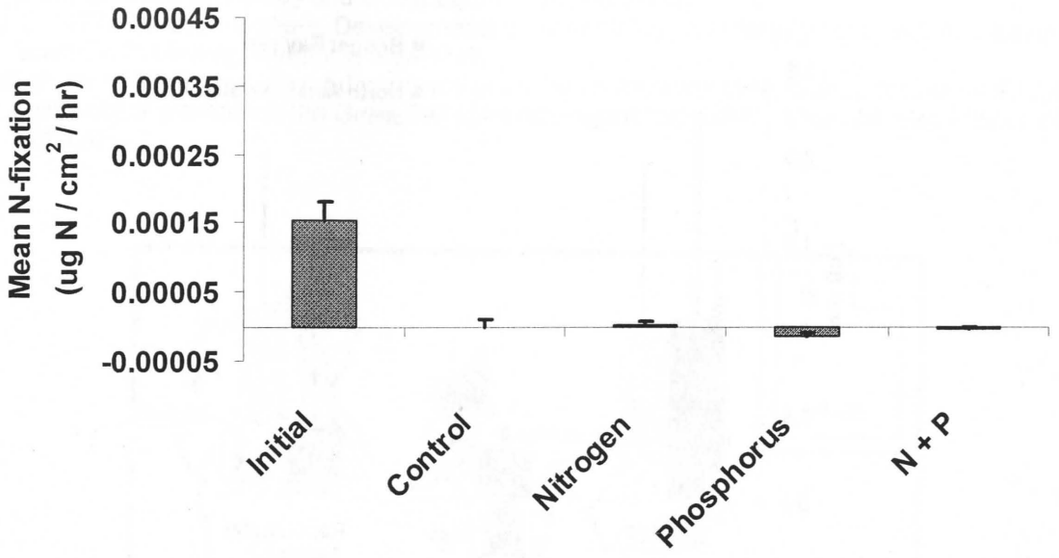

Figure 4. Bars represent the mean nitrogen fixation rate of stromatolites from the initial samples and at the end of the nutrient addition bioassay. Error bars represent +1 standard error. It is important to note that although initial nitrogen fixation appears to be significantly larger than other treatments, the actual rate is miniscule and can be attributed to laboratory error.

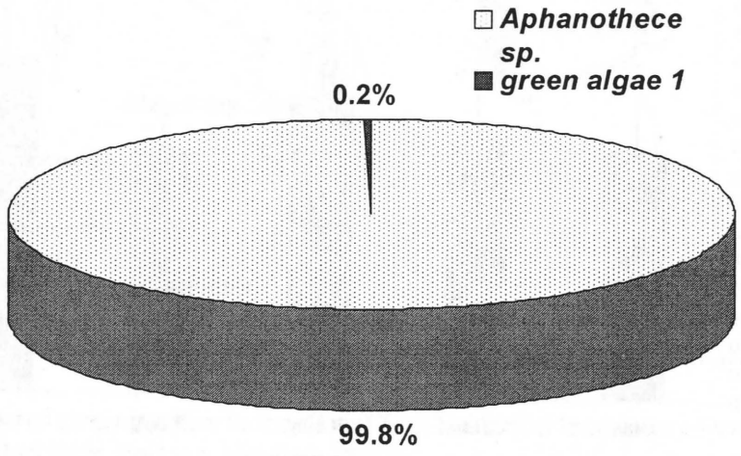

Figure 5. Relative abundance of taxa found on stromatolites from the Great Salt Lake. 513 individuals of *Aphanothece* sp. were found while only one green algae individual was found.

Tables

Table 1. Length, width, height, area, and shape of all stromatolite samples at both sampling locations

Location	Sample number	Length (m)	Width (m)	Depth to crest (m)	Depth to surrounding substrate (m)	stromatolite height (m)	Approximate shape	area (m^2)
North Antelope Island	1	1.54	0.95	0.85	0.98	0.14	N/A	N/A
North Antelope Island	2	0.75	0.75	0.82	0.96	0.14	N/A	N/A
North Antelope Island	3	0.94	0.90	0.81	0.98	0.16	N/A	N/A
North Antelope Island	4	0.55	0.40	0.93	0.98	0.05	N/A	N/A
North Antelope Island	5	0.40	0.36	0.90	0.98	0.08	N/A	N/A
North Antelope Island	6	0.80	0.66	0.88	0.99	0.11	N/A	N/A
North Antelope Island	7	1.40	0.84	0.83	0.97	0.15	N/A	N/A
North Antelope Island	8	0.63	0.60	0.88	0.99	0.12	N/A	N/A
North Antelope Island	9	1.50	0.80	0.88	0.90	0.02	elliptical	0.94
North Antelope Island	10	1.00	0.80	0.79	0.95	0.16	circular	0.64
North Antelope Island	11	0.90	0.60	0.81	0.99	0.18	elliptical	0.42
North Antelope Island	12	0.91	0.75	0.90	0.98	0.08	circular	0.54
North Antelope Island	13	1.20	0.50	0.92	0.99	0.06	rectangular	0.60
North Antelope Island	14	0.88	0.39	0.86	0.93	0.07	elliptical	0.27
North Antelope Island	15	1.40	0.80	0.88	0.98	0.11	elliptical	0.88
North Antelope Island	16	1.50	0.80	0.79	0.99	0.20	elliptical	0.94
North Antelope Island	17	1.00	0.90	0.80	0.92	0.12	circular	0.71
North Antelope Island	18	1.60	1.20	0.78	0.94	0.16	triangular	0.66
North Antelope Island	19	2.30	1.80	0.74	0.91	0.17	circular	3.30
North Antelope Island	20	0.86	0.76	0.80	0.91	0.11	circular	0.52
North Antelope Island	21	0.95	0.89	0.82	0.95	0.13	circular	0.66
North Antelope Island	22	1.10	0.85	0.83	0.94	0.11	elliptical	0.73
North Antelope Island	23	1.30	0.87	0.69	0.78	0.09	circular	0.92
North Antelope Island	24	0.80	0.78	0.76	0.84	0.08	rectangular	0.62
North Antelope Island	25	1.30	0.77	0.77	0.92	0.15	elliptical	0.79
North Antelope Island	26	1.00	0.73	0.77	0.80	0.03	rectangular	0.73
North Antelope Island	27	1.10	0.85	0.79	0.90	0.11	elliptical	0.73
North Antelope Island	28	2.40	1.00	0.81	0.93	0.12	rectangular	2.40
North Antelope Island	29	1.10	0.90	0.79	0.90	0.11	circular	0.79
North Antelope Island	30	2.70	1.20	N/A	N/A	0.10	N/A	N/A
North Antelope Island	31	1.50	1.00	N/A	N/A	0.20	N/A	N/A
North Antelope Island	32	1.00	1.00	N/A	N/A	0.14	N/A	N/A
North Antelope Island	33	1.10	0.60	N/A	N/A	0.17	N/A	N/A
North Antelope Island	34	1.50	0.80	N/A	N/A	0.22	N/A	N/A
North Antelope Island	35	1.50	0.70	N/A	N/A	0.12	N/A	N/A
North Antelope Island	36	2.50	0.80	N/A	N/A	0.14	N/A	N/A
North Antelope Island	37	1.60	0.60	N/A	N/A	0.21	N/A	N/A
North Antelope Island	38	0.80	0.80	N/A	N/A	0.20	N/A	N/A
North Antelope Island	39	1.00	0.80	N/A	N/A	0.20	N/A	N/A
North Antelope Island	40	1.00	0.70	N/A	N/A	0.13	N/A	N/A
mean values		1.23	0.81			0.13		0.90
standard deviations		0.52	0.24			0.05		0.69

Bridger Bay	41	0.80	0.45	0.53	0.61	0.09	elliptical	0.28
Bridger Bay	42	1.00	0.65	0.48	0.64	0.16	elliptical	0.51
Bridger Bay	43	0.75	0.50	0.49	0.65	0.16	elliptical	0.29
Bridger Bay	44	0.30	0.35	0.58	0.64	0.06	heart	N/A
Bridger Bay	45	0.38	0.30	0.59	0.63	0.04	triangular	N/A
Bridger Bay	46	0.45	0.15	0.55	0.65	0.10	triangular	N/A
Bridger Bay	47	2.40	1.20	0.58	0.62	0.04	circular	2.54
Bridger Bay	48	0.70	0.65	0.57	0.67	0.10	circular	0.36
Bridger Bay	49	0.55	0.25	0.63	0.65	0.02	elliptical	0.11
Bridger Bay	50	0.50	0.30	0.62	0.67	0.05	rectangular	0.15
Bridger Bay	51	0.60	0.58	0.55	0.66	0.11	circular	0.27
Bridger Bay	52	0.95	0.55	0.54	0.66	0.12	elliptical	0.41
Bridger Bay	53	0.70	0.58	0.53	0.66	0.13	rectangular	0.41
Bridger Bay	54	0.85	0.75	0.57	0.63	0.06	rectangular	0.64
Bridger Bay	55	0.60	0.22	0.61	0.68	0.07	elliptical	0.10
Bridger Bay	56	0.32	0.22	0.59	0.66	0.07	rectangular	0.07
Bridger Bay	57	0.91	0.85	0.52	0.58	0.06	rectangular	0.77
Bridger Bay	58	1.10	0.49	0.49	0.61	0.12	elliptical	0.42
Bridger Bay	59	0.85	0.50	0.52	0.63	0.11	elliptical	0.33
Bridger Bay	60	0.55	0.27	0.57	0.64	0.07	elliptical	0.12
mean values		0.76	0.49			0.09		0.46
standard deviations		0.45	0.25			0.04		0.57

Table 2. As a means of testing the potential statistical differences between treatments a single-factor ANOVA was run using the chlorophyll α concentrations of each treatment. Note that there are five replicate samples for each treatment and that there is no statistical difference.

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Control	5	231.40	46.28	547.94
Nitrogen	5	265.31	53.06	469.32
Phosphorus	5	215.84	43.17	177.98
Nitrogen & Phosphorus	5	295.01	59.00	501.10

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	751.82	3	250.61	0.591	**0.630**	**3.239**
Within Groups	6785.36	16	424.08			
Total	7537.18	19				

Table 3. Diameters of *Aphanothece* sp. as measured in three sub-samples. Note that samples 1-10 comprise Sample A; 11-20 comprise Sample B; and 21-30 comprise Sample C. Sample 31 – in italics – represents the one species of green algae that was found.

sample number	Diameter (micrometer units)	Diameter (um)
1	2	1
2	3	2
3	2.5	2
4	3.5	2
5	3	2
6	1	1
7	1.5	1
8	1.5	1
9	1.5	1
10	2.5	2
11	2.5	2
12	1	1
13	2	1
14	2.5	2
15	1.5	1
16	3	2
17	1.5	1
18	2.5	2
19	2	1
20	2.5	2
21	1.5	1
22	1	1
23	2	1
24	2.5	2
25	1.5	1
26	2	1
27	2	1
28	1	1
29	3	2
30	2	1
31	*9*	*23*

Chapter 4

Cryptobiology, Paleolimnology and Selenium Sensitivity of Brine Shrimp from the Great Salt Lake, Utah, USA

Summary

The objective of this study was to investigate the presence and viability of brine shrimp (*Artemia franciscana*) cysts stored as an egg bank in the sediments of Gilbert Bay, Great Salt Lake. We were interested in determining if recently deposited cysts were more resistant to selenium than those deposited prior to the settlement by Anglos in the Salt Lake Valley in 1869. We used a gravity corer to collect a 43-cm long sediment core from Gilbert Bay. We then sectioned the core in 1-cm intervals, froze the samples for 21 d to simulate extreme environmental conditions, and separated brine shrimp cysts from the sediments with saturated salt water. The cysts were then hatched in 3.5% NaCl. We successfully hatched between 1 and 20 cysts from each section down to a depth of 25-26 cm. With expected sedimentation rates of 0.71 mm yr^{-1}, we estimate the deepest hatched cysts to be approximately 360 years old.

With the low number of brine shrimp nauplii hatched from each section, we could not compare the sensitivity of recent and ancient genotypes to selenium. Instead, a bioassay was conducted to measure the lethal concentration of selenium for brine shrimp nauplii hatched from commercially-obtained cysts from the Great Salt Lake. The assay was conducted in 15% NaCl solution at 20°C. The 48-hr LC_{50} was 27 mg Se L^{-1}, and the 96-hr LC_{50} for newly-hatched nauplii was <<30 mg Se L^{-1}, the lowest concentration tested. These levels are considerably less than site-specific toxicity levels described by Brix et al. (2004), suggesting that a more careful analysis of selenium toxicity to brine shrimp is needed.

Our work will aid scientists and future aquatic practicum classes at USU in their continuing studies of the Great Salt Lake. The study demonstrates the possibility of using prehistoric cysts of brine shrimp to determine if brine shrimp have adapted to changing environmental conditions in the Great Salt Lake ecosystem.

Kathleen B. Markland
Department of Aquatic Watershed and Earth Resources
Utah State University

Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 Dec 2004

Introduction

The Great Salt Lake is dependent on climatic events for its influx of water and salts from the Wasatch Mountains. The balance between the influx from spring runoff and the evaporation rate in the summer determines the water surface elevation of the lake. When a river used for irrigation and/or copper processing plants, such as the Jordan River, feds a terminal lake, the evaporation process concentrates metals in the water, potentially leading to toxic levels of contaminants (Dodds 2002). Copper mining and smelting has been a major industry in the Salt Lake region since the late 1800's. Kennecott Utah Copper Company has operated an open pit copper mine in the Oquirrh Mountains (Stokes 1986) located in southwest Salt Lake Valley and presently utilizes a discharge outlet into Gilbert Bay of the Great Salt Lake (Brix 2004). In addition to copper, a variety of other metal contaminants are produced in the smelting process, many of which ultimately wind up in the Great Salt Lake. Contaminant accumulation in the Great Salt Lake since Anglo settlement has been documented by the US Geological Survey who analyzed a 24-cm long sediment core sample from Farmington Bay. Their analysis demonstrated conclusively that contaminated sediments have been deposited, most likely from anthropogenic causes, since the early to mid 1900's. The contamination has continued to increase, especially from 1979 to 1998 (Naftz et al. 2000).

Selenium is one contaminant of concern in the Great Salt Lake that is produced by mining activities. In 2001, the US Geological Survey collected water-column samples from the lake at 13 sites and found selenium levels to range between 20 and 60 $\mu g\ L^{-1}$ (Waddell et al. 2002). In contrast, the maximum allowable concentration of selenium in drinking water is 10 $\mu g\ L^{-1}$ (Dodds 2002). This criterion does not apply to a saline lake with a hardness level greater than 50 $mg\ L^{-1}$ and high concentrations of sulfate, but helps to display the high level of selenium in the Great Salt Lake. Selenium concentrations of 300 $\mu g\ L^{-1}$ have been recorded near the Kennecott outfall discharge in Gilbert Bay (Brix et al. 2004). The industry is currently discussing increasing discharges of selenium into the Great Salt Lake, and there is concern that it might bioaccumulate in the food web (Lemly 1997). The ecosystem of the Great Salt Lake is somewhat simplified, with a limited number of phytoplankton and periphyton taxa, and only two abundant invertebrates, brine shrimp and brine flies. Because the system lacks fish, the selenium passed up the food web to the brine shrimp and brine flies can bioaccumulate directly to birds that feed on them. For example, eared grebes feed extensively on brine shrimp in the Great Salt Lake before continuing their migration path. The US Fish and Wildlife Service conducted a preliminary survey in the lake and in a limited sampling found selenium levels to nearly double from 6.2 to 10.9 µg per g dry weight in eared grebe livers between late September and early December (Waddell et al. 2002).

The physiology and life history of brine shrimp is adapted to extreme natural environmental changes, and they may also adapt to anthropogenic stressors. They can reproduce quickly producing live nauplii under optimal conditions. This process is called ovoviparity. When conditions become unfavorable, the brine shrimp have the flexibility to quickly switch to producing cysts, a process termed oviparity. For instance, in the fall when the temperature cools, they produce cysts that float on the surface of the water. A multi-million dollar industry has been created harvesting the cysts. The cysts that remain can mix seasonally with the water column. At lower salinities the cysts loose buoyancy and descend to the sediments where they become buried. Unhatched, these buried cysts serve as an egg bank. If the species is extirpated in the lake, the buried cysts can repopulate the ecosystem when optimal conditions return. This process serves as temporal dispersal, because in a fragmented habitat, such as a terminal lake basin, spatial dispersal may not be an option (Hairston 2002). The cysts stay in a cryptobiotic state until their diapause is broken by a change in environmental conditions which cue the hatching process to begin. One study found the diapausing eggs of other zooplankton to cue on oxygen levels and light (Hairston 2002). Manuals for hatching brine

shrimp cysts for aquaculture, however, suggest that cysts should be frozen and dehydrated to break diapause (Lavens and Sorgeloos 1987). This process occurs naturally in the Great Salt Lake where winter water temperatures drop below 0°C and the high salt concentration can dehydrate the cysts. However, it is likely that cysts have to be within one millimeter of the top of the sediment to be able to reach the pelagic zone once they hatch (Hairston 2002).

Detritus is continually descending through the water column and settling on the bottom in a lake ecosystem. This detritus leaves a layered history of the past ecosystem that limnologists reconstruct using pigment analysis, isotope analysis or extreme environmental events. For instance, the Mount Mazama eruption 6,800 years ago deposited an ash layer in the Great Salt Lake and now lies at a depth of 485 cm in the sediments. This indicates that sedimentation rates in Gilbert Bay have averaged 0.71 mm yr^{-1} since the eruption (R. Thompson, USGS, pers. comm.). In contrast, David Naftz (pers. comm.) estimated the sedimentation rate in Farmington Bay to be 1 mm yr^{-1} because of higher sediment delivery rates from the Jordan River.

The influx of water and solutes to the lake varies with climatic change and anthropogenic diversions, but pollution, including selenium, has been increasing since 1979 (Naftz et al. 2000; Brix 2004). Presently, a narrative criterion is used for water quality standards in the Great Salt Lake. The standards for a hypersaline lake must be different than for a freshwater lake, and recent collaborative efforts to set numerical standards, beginning with selenium, require repetitive studies with good experimental design. The objective of our study was to compare the sensitivity of recently deposited cysts to selenium with those deposited prior to the settlement of Anglos in the Salt Lake Valley.

Methods

Study Area–The Great Salt Lake is adjacent to the metropolitan area of the Wasatch Front in northern Utah. Nearly 2 million people live within the Great Salt Lake Basin (U.S. Census Bureau 2003) between the large shallow hypersaline terminal lake and the snow-dominated Wasatch Mountains. The Great Salt Lake is the shrunken remnant of the larger, freshwater, ancient Lake Bonneville (Stokes 1986). Typical of a terminal lake, there is a delicate balance between influx and evaporation; the region is presently in the sixth year of a drought. In addition, numerous causeways have divided the lake into sections that prevent mixing, thus creating high concentrations of salt and pollutants. We collected our sediment cores in Gilbert Bay (41 04.226 N 112 16.089 W) on September 30, 2004. At collection, the water depth at the site was 6.5 meters and the mean salinity was 17%. The Secchi depth was 3.1 meters, and the temperature in the unstratified water column was 18°C.

Field sampling and core processing–We used a gravity-corer (K.B. Type 3 SN 095, 2.500 Core, JR Glew Mfg) with a 50-cm long tube to collect three cores. The core sampler was hand suspended vertically off the back platform of the USU research boat, dropped to about 1 m above the sediment surface, then allowed to fall under its own weight into the sediments. A bronze messenger sent down the rope triggered a plunger mechanism. We retrieved the core and as it cleared the water surface, the bottom plug was inserted. The sediment core was disconnected from the gravity apparatus, placed vertically in a five gallon plastic bucket with packing foam, and covered with garbage bags to exclude light (Leavitt 2001). Care was taken to keep the samples vertical during transport. The next morning at 10:00 the core samples were placed in a dark incubation chamber and kept at a constant temperature of 10 ± 1°C until the 2 of the 3 cores were sectioned 6 and 16 days later.

Cores were sectioned into discrete intervals. We used the Glew core sectioning apparatus to slice the core #1 at 5-mm intervals. Extreme care was taken to isolate each core interval, with a thorough washing with distilled water of the slicing tray and tools between each increment. We did not, however, remove the outside edges of each section, so it is possible

that during the coring process, some surficial sediments were pushed along the outside edge of the core by the plastic tube. Sections from Core #1 were split, half of each section was stored in rinsed, black film canisters and placed vertically in cold storage at 0 – 4 ° C (Leavitt 2001). Dr. Peter Leavitt at the University of Regina, Canada is presently performing a pigment and stable isotope analysis on these sections and will date representative sections. After 15 days, core #2 was sectioned at 10-mm intervals and stored at 4ºC for 21 days prior to further treatment. Core #3 has not yet been sectioned and will provide material for future work.

Incubation of brine shrimp cyst–The second half of core #1 was saved in one centimeter vertical segments, placed in preweighed crucibles and dried in an oven at ca. 24° C. I carefully stirred the sediment to increase the surface area and decrease the drying time (Lavens and Sorgeloos 1987). Unfortunately, the oven malfunctioned, temperatures increased to above 40ºC, and we considered the samples lost.

The sediment sections from core #2 were consequently thawed overnight and placed in 250-ml flasks containing 100 ml of 30% sodium chloride solution for three days. During this period they were kept in the dark at a temperature of 10° C. This helped to break diapause (Lavens and Sorgeloos 1987) and floated the cysts out of the sediments to the surface. To hatch the cysts, they were decanted into 450 ml plastic jars containing distilled water with 3.5% reagent grade sodium chloride. The jars were randomly placed in an incubation room under constant light (120 µE m^{-2} s^{-1}) and at a temperature of 25 ± 1° C for the duration of the incubation. The number of nauplii that hatched in each jar was observed after 1, 3, 6, 8 and 11 days. The highest number of nauplii observed on any of these days is presented. After hatching, the nauplii were fed a phytoplankton mixture obtained from a 1:1 mix of Gilbert Bay and Farmington Bay water that was filtered through an 80 µm sieve to remove large plankton.

Selenium bioassay–Commercial cysts from Saunders Brine Shrimp, Ogden, UT, that were purchased in 2002 and stored in a refrigerator were used to produce nauplii. They were hatched in 3.5% sodium chloride in an aerated glass cone at 29 ± 1° C. The cysts hatched in ca. 18 hr.

A 20 g Se L^{-1} stock solution[1] of sodium selenite (CAS 10102-18-8) was prepared in a 500-ml volumetric flask. Eight test solutions, with two replicates each, were prepared: 0, 30, 60, 90, 120, 150, 180 and 210 mg Se L^{-1}, based on the toxicity levels found by Brix et al. (2004). The solutions were prepared with a mixture of 600 ml of 80 µm-filtered Farmington Bay water (3.5%) and 2400 ml 15% reagent grade sodium chloride solution to give a final salinity of 12.8%. The Farmington Bay water was used to provide a food source for the nauplii. In sequence, the calculated stock solution was placed in a 500-ml volumetric flask with a pipette and was diluted to 500 ml with the mixture described above. 200 ml was measured in graduated cylinder and poured into well rinsed and labeled 250-mL Erlenmeyer flasks.

Twenty nauplii < 24 hours old were randomly placed in each test solution and incubated with 24-hr lighting (120 µE m^{-2} s^{-1}) at 25 ± 1° C. Surviving nauplii were counted after 24, 48, 72, and 96 hours. The numbers of surviving nauplii were normalized to the control group and the percent mortality was calculated and presented as "corrected mortality" which equals:

$$\text{Corrected Mortality (\%)} = \frac{(\text{\# dead}) - (\text{mean \# dead in control}) * 100}{20 - (\text{\# dead in control})}$$

Lethal concentrations to 50% of the control-normalized population (LC_{50}) were determined by graphical analyses.

---

[1] Stock solution: sodium selenite (powder-dry) hydrated produces selenious acid.
(20g/L)*(173g H_2SeO_3)/(79g Se) = 43.8g Sodium Selenite/L = 21.9 g / 500 ml

Results

Brine shrimp hatching from core–A visual assessment of the core found that the top 25 cm was dark, probably indicating a high proportion of organic matter. Below this depth the sediment was primarily light grey with little dark material.

Ninety percent of the cysts hatched within 2 days of being placed in 3.5% salinity water, but some additional nauplii continued to hatch until the end of the 10-day observation period (Table 1). The highest hatch rates occurred in the top 9 cm of the core, with a mean rate of 17 nauplii produced per section (Figure 1). However, brine shrimp cysts hatched from the core down to a depth of 25-26 cm. At this depth, with a sedimentation rate of 0.71 mm yr^{-1}, the cysts were estimated to be 360 years old.

Insufficient cysts hatched from each section to perform a bioassay, because 160 individuals would have been needed from each section tested. Instead, the brine shrimp are being reared in the laboratory and their progeny may be used for future bioassay experiments.

Selenium bioassay–At 96 hr, all of the nauplii were dead in the lowest selenium concentration tested (30 mg L^{-1}), making it impossible to determine the LC_{50} at that time interval (Table 2; Figure 2). Mortality in the control treatments was substantial with 38% dying after 48 hours and 55% by 96 hours (Table 3). However, the variability between control treatments was relatively low, particularly at 48-hr, and the LC_{50} was estimated to be 27 mg Se L^{-1} (Figure 3).

Discussion

We successfully hatched brine shrimp (*Artemia franciscana*) cysts found 26 cm deep in Great Salt Lake sediment. The dark greenish tinge in the sediment ended about there. Based on the sedimentation rate of 0.71 mm yr^{-1}, we calculate the oldest cysts to be approximately 360 years old. Therefore, we concluded that the viability of the cysts last about 350 years or longer in this environment. It will be necessary to date actual core material before this longevity can be verified, rather than relying on a mean sedimentation rate determined over a much longer interval. It is possible that the light grey sediments dominant below 25 cm indicated that our sample site was not overlain by a lake at that time, as dark organic material is preserved well in anoxic sediments, but poorly in oxidized surface soils. It would be interesting to obtain cores from deeper area of the lake where continuous organic-rich sediments were present. Conceivably, viable cysts might then be found even deeper in the sediments. It is also possible that brine shrimp disappeared from the lake at certain times when salinity levels dropped and allowed invertebrate and fish predators to occupy the Great Salt Lake (Wurtsbaugh 1992). Analysis of algal pigments from the core is currently underway, and it may be possible to determine salinity levels from the algal remains left at different depths in the sediments.

The methodology employed for dehydrating and removing the cysts from the sediments with saturated salt water worked well. However, some cysts were lost when the brine was decanted. Another methodological improvement would be to more carefully control the algae that the nauplii are fed. Substantial mortality of the hatched nauplii occurred, and may of those that initially hatched died within 15 days.

Paleolimnologists have used sedimented remains of algae and zooplankton to provide valuable reconstructions of past environmental conditions. Our results demonstrate that brine shrimp cysts that are several hundred years old can be hatched to provide cryptobiotic information about prehistoric conditions in the Great Salt Lake, as has been done with resting eggs of zooplankton from other lake environments (Hairston et al. 1999). The genotype and phenotype of *Artemia* could be studied. Perhaps we could discover more about their adaptive ability and how their sensitivity to chemicals in the ecosystem changes over time. Overall, it was very exciting breaking the brine shrimp diapause, especially of eggs that may have been

deposited over 300 years ago. People are fascinated with the concept of cryptobiosis and the approach provides great potential for future scientific research.

Our bioassay indicated that brine shrimp nauplii may be more sensitive to selenium than was reported by Brix et al. (2004), as they reported a 96-hr LC_{50} of 71-86 mg / L, three times the concentration we estimated for a 48-hr LC_{50}. There are at least three possible reasons for this difference. First, the source of brine shrimp used by Brix et al. was not reported, and they may have used cysts obtained from Farmington Bay (W. Moelmer, Utah Division of Water Quality, pers. comm.). Because local adaptations may be possible, toxicity tests should be done on shrimp from the Gilbert Bay, like those used in our study. Secondly, it is unclear how Brix et al. (2004) reported their concentrations. In their introduction they state that "unless otherwise noted, all discussion of Se in this paper is referring to selenate", but subsequently in the paper they appear to give concentrations as Se (our emphases). If they were actually reporting concentrations of selenate, the results of the two bioassays do not differ that much. A third major difference between the two assays was that our test water contained relatively little sulfate compared to the water used by Brix et al. (2004). Because sulfate is a competitive inhibitor of selenium uptake (Forsythe and Klaine 1994), it is likely that nauplii in our assay were more susceptible to the metal than in Brix et al's bioassay. Finally, the nauplii in our control treatments had high mortality, which compromises the interpretation of our estimated LC_{50}. Nevertheless, it is clear from our results that the 96-hr LC_{50} was <30 mg L^{-1}, as no shrimp survived at that concentration. Given all of these issues, it is clear that additional assays are needed to resolve the sensitivity of brine shrimp in the Great Salt Lake to selenium.

Acknowledgments

We thank Peter Leavitt from the University of Regina, who provided valuable advice at the onset of the project. Wim Tackaert from INVE, Inc. recommended using brine to separate cysts from the sediments and to dehydrate them. Mike Pfrender from Utah State University provided the corer and guidance on sectioning the core. Robert Thompson, Walter Dean and David Naftz from the U.S. Geological Survey provided sedimentation rates for the Great Salt Lake and advice on interpreting our results.

References

Brix KV, Deforest DK, Cardwell RD, Adams WJ. 2004. Derivation of a chronic site-specific water quality standard for selenium in the Great Salt Lake, Utah, USA. Environmental Toxicology and Chemistry. 23: 606-612.

Dodds WK. 2002. Freshwater Ecology: Concepts& Environmental Applications. Academic Press.

Forsythe BL, Klaine SJ. 1994. The interaction of sulfate and selenate effects on brine shrimp, *Artemia* spp.. Chemosphere 29: 789-800.

Hairston NG, Lampert W, Caceres CE, Holtmeier CL, Weider LJ, Gaedke U, Fischer JM, Fox JA, Post DM. 1999. Lake ecosystems - Rapid evolution revealed by dormant eggs. Nature 401: 446-446.

Hairston NG, Kearns CM. 2002. Temporal dispersal: ecological and evolutionary aspects of zooplankton egg banks and the role of sediment mixing. Integrative and comparative biology 42: 481-491

Lavins P, Sorgeloos P. 1987. The cryptobiotic state of Artemia cysts, its diapause deactivation and hatching: a review. *Artemia* research and its applications 3: 27-63.

Leavitt PR, Hodgson DA. 2001. Sedimentary pigments. Pp 295-325 In: Smol JP, Birks HJB and Last WM (eds.), Tracking Environmental Change using Lake Sediments. Volume 3: Terrestrial, Algal and Siliceous Indicators. Kluwer.

Lemly DA. 1997. Environmental Implications of Excessive Selenium: A Review. Biomedical and Environmental Sciences 10: 415-435.

Naftz DL, Stephens DW, Callender E, Van Metre PC. 2000. Reconstructing historical changes in the environmental health of watersheds by using sediment cores from lakes and reservoirs in Salt Lake Valley, Utah. USGS FS–164-00. (http://ut.water.usgs.gov)

U.S. Census Bureau, Census data 2000 for the State of Utah, Nov.24, 2004, fact finder.census.gov.
Stokes LE. 1986. Geology of Utah. Utah Museum of Natural History.
Wurtsbaugh WA. 1992. Food-web modification by an invertebrate predator in the Great Salt Lake (USA). Oecologia 89:168-75.
Waddell B, Naftz DL, Kimball BA and Garbarino JR. 2002 (abstract). Preliminary investigation of selenium in water, bottom sediment and biota, Great Salt Lake, Utah. SETAC meeting presentation.

Tables

Table 1. Brine shrimp nauplii hatched from different strata of a sediment core taken from Gilbert Bay in the Great Salt Lake. Cysts were placed in hatching solution on 9 Nov 2004, and observed on five subsequent dates. The estimate age of the cysts and nauplii and the deposition date is based on an assumed sedimentation rate of 0.71 mm year^{-1}.

Core Depth (cm)	10-Nov-04	12-Nov-04	15-Nov-04	17-Nov-04	20-Nov-04	Maximum observed	Estimated age (yr)	Estimated Deposition Date
0-1	7	20	0	0	0	20	7	1997
1-2	2	11	3	1	0	11	21	1983
2-3	5	17	3	0	0	17	35	1969
3-4	6	11	9	8	9	11	49	1955
4-5	4	17	14	15	19	19	63	1941
5-6	7	15	12	14	20	20	77	1927
6-7	9	18	20	19	20	20	92	1912
7-8	10	14	6	3	2	14	106	1898
8-9	12	18	13	13	13	18	120	1884
9-10	4	11	11	10	11	11	134	1870
10-11	1	6	7	10	10	10	148	1856
11-12	2	2	3	3	3	3	162	1842
12-13	2	10	9	14	14	14	176	1828
13-14	2	13	6	10	10	13	190	1814
14-15	1	8	8	12	14	14	204	1800
15-16	1	5	3	5	5	5	218	1786
16-17	4	4	1	2	2	4	232	1772
17-18	0	5	3	3	4	5	246	1758
18-19	0	2	0	1	2	2	261	1743
19-20	1	5	3	6	4	6	275	1729
20-21	2	4	2	0	0	4	289	1715
21-22	1	4	0	0	0	4	303	1701
22-23	0	2	1	1	2	2	317	1687
23-24	0	4	0	2	0	4	331	1673
24-25	0	1	0	0	0	1	345	1659
25-26	0	0	0	1	1	1	359	1645
26-27	0	0	0	0	0	0	373	1631
27-28	0	0	0	0	0	0	387	1617
28-29	0	0	0	0	0	0	401	1603
29-30	0	0	0	0	0	0	415	1589
31-32	0	0	0	0	0	0	430	1574
32-33	0	0	0	0	0	0	444	1560
33-34	0	0	0	0	0	0	458	1546
34-35	0	0	0	0	0	0	472	1532
35-36	0	0	0	0	0	0	486	1518
36-37	0	0	0	0	0	0	500	1504
37-38	0	0	0	0	0	0	514	1490
38-39	0	0	0	0	0	0	528	1476
39-40	0	0	0	0	0	0	542	1462
41-42	0	0	0	0	0	0	556	1448
42-43	0	0	0	0	0	0	570	1434
Total Core	83	227	137	153	165	253		
% of Max	33%	90%	54%	60%	65%	100%		

Table 2. The mean number of dead *Artemia franciscana* nauplii counted at 24 hour intervals in 7 selenium concentrations and the control group. All samples began with 20 nauplii less then 24 hours old. Each concentration had two replicates and these counts were averaged.

Se conc.(mg L^{-1})	Start	24 hours	48 hours	72 hours	96 hours
0	0	7	7.5	10	11
30	0	8	14.5	15	20
60	0	10.5	16.5	17	20
90	0	17.5	18	20	20
120	0	19	19	20	20
150	0	16.5	20	20	20
180	0	20	20	20	20
210	0	20	20	20	20

Table 3. Mean corrected Mortality (%) of nauplii in 7 concentrations of selenium and in the control treatments

Se conc.(mg L^{-1})	start	24 hours	48 hours	72 hours	96 hours
0	0				
30	0	7.7	56	50	100
60	0	26.9	72	65	100
90	0	80.8	84	100	100
120	0	92.3	92	100	100
150	0	100	100	100	100
180	0	100	100	100	100
210	0	100	100	100	100

Figures

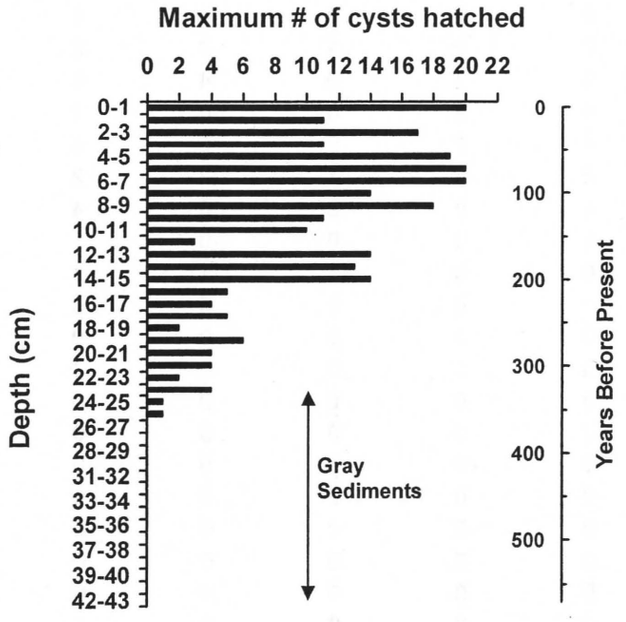

Figure 1. Numbers of brine shrimp (*Artemia franciscana)* that hatched from different strata of a 43-cm long sediment core taken in the Great Salt Lake, Utah. A constant sedimentation rate of 0.71 mm $year^{-1}$ was assumed in order to estimate the age of each core section.

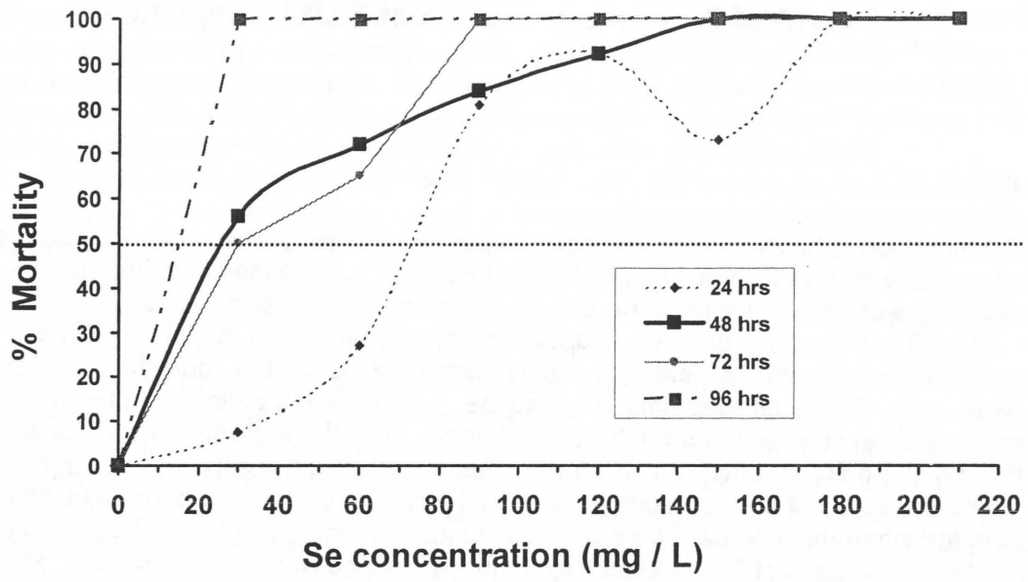

Figure 2. Percent mortality of brine shrimp (*Artemia franciscana*) nauplii exposed to varying selenium concentrations. They were counted every 24 hours for the total duration of 96 hours.

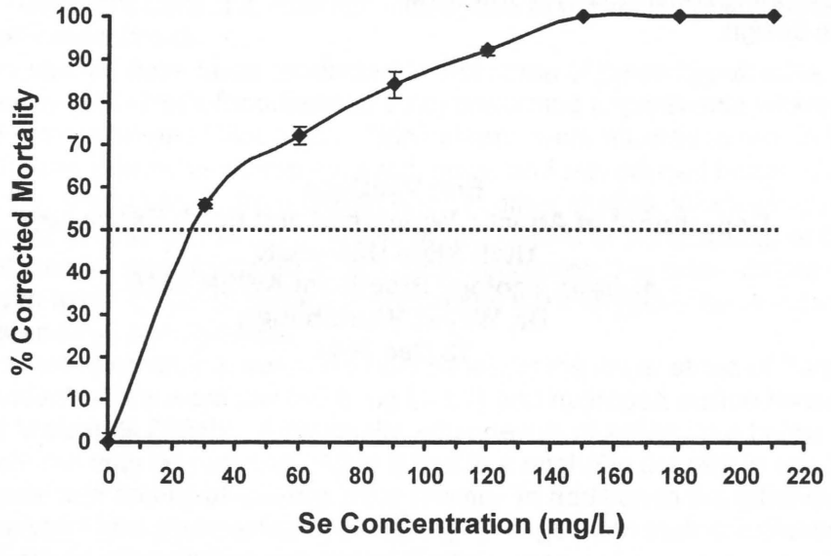

Figure 3. Mortality of brine shrimp nauplii exposed to different concentrations of selenium after 48 hours. The estimated 48hr LC_{50} for the Se was 27 mg L^{-1}. Error bars, when they extend beyond the data point, show the standard deviations of the two replicate flasks tested for each concentration.

Chapter 5

Survival of Artemia in the Great Salt Lake, Utah

Summary

It is not entirely known why the densities of brine shrimp (*Artemia franciscana)* are lower in Farmington Bay than in Gilbert Bay of the Great Salt Lake. To assess the importance of water quality as a control factor for brine shrimp, nauplii were placed in meshed cages at depths of 0.2 m and 0.8 m in both Farmington and Gilbert bays for one week in October 2004. Numerous macrozooplankton entered the cages during the incubation, indicating that the cages had leaks that could have allowed nauplii to escape, thus making it difficult to interpret results. However, the field experiment demonstrated that nearly all of the extraneous corixids and harpacticoid copepods that entered the 0.8-m cages died during the experiment, suggesting that water quality there did not support zooplankton. A 1-week laboratory experiment using lake water collected when the field cages were deployed also demonstrated that nauplii could not survive in the 0.8 m water of Farmington Bay, or in the surface water of Gilbert Bay. Mean H_2S concentrations of 20 mg L^{-1} and anoxia in the 0.8 m water from Farmington Bay likely killed the nauplii, and low phytoplankton levels in the Gilbert Bay water may have contributed to mortality in that water. The survival of some nauplii in the field cages indicates that this approach has promise for evaluating water quality in the Great Salt Lake, but additional work will be necessary to optimize the design.

Erin VanDyke
Department of Aquatic Watershed and Earth Resources
Utah State University

Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 Dec 2004

Introduction

Brine shrimp, *Artemia franciscana*, are very abundant in Gilbert Bay of the Great Salt Lake, but usually absent in the less saline and hypereutrophic Farmington Bay. However, the reason for this difference is not know. The focus of my project was to develop a methodology for testing survival of the brine shrimp under natural conditions in the Great Salt Lake and to determine survival rates of nauplii in both bays for one week in October, 2004

Farmington and Gilbert Bays of the Great Salt Lake are separated by a causeway that joins the Wasatch Front with Antelope Island, and very different conditions prevail in the two bays. Farmington Bay is located at the southern end on the Great Salt Lake and is considered hypereutrophic because of high nutrient loading (Wurtsbaugh and Marcarelli 2004a). Farmington Bay is sometimes characterized by much lower salinities (~ 3.3%) than Gilbert Bay (~17%). Gilbert Bay is not as eutrophic as Farmington Bay; however, there is a breech in the causeway that allows Farmington Bay water to move into Gilbert Bay. This movement leads to a plume of nutrients that extends into Gilbert Bay (see chapter 2 of this report by J. Robinson).

Brine shrimp are an important food resource for migrating waterfowl and the cysts of brine shrimp are an important commercial resource (Nicholson and Marcarelli 2004). Brine shrimp occur at higher densities in Gilbert Bay than in Farmington Bay for most of the year (Wurtsbaugh and Marcarelli 2004a). It is not completely know why this occurs. Wurtsbaugh and Marcarelli (2004a) state that the causes could include: anoxia from high nutrient loading, low salinity, predation by corixids and predatory copepods, algae taxa that are toxic to brine shrimp, and possibly hydrogen sulfide from the sediments that could mix throughout the water column during high wind events. The current knowledge of brine shrimp survival under natural conditions in the Great Salt Lake is somewhat limited, due to difficulties encountered in the application of *in situ* experiments.

Laboratory studies have been conducted to test some of these hypotheses. For instance, Wurtsbaugh and Gross (unpublished data) preformed experiments where water was taken from both Farmington and Gilbert Bay. Brine shrimp were allowed to rear in that water and the results showed that brine shrimp survived, grew, and reproduced better in the water from Farmington Bay than in water from Gilbert Bay. In other studies, Wurtsbaugh et al. (2002) showed that Farmington Bay can become anoxic at early hours in the morning, or for prolonged periods after wind storms, and Marcarelli et al. (2003) suggested that brine shrimp could avoid anoxia by moving to water closer to surface that contains more oxygen. More research is needed to understand this phenomenon.

I predicted that brine shrimp survival would be low in the lower strata of Farmington Bay where oxygen concentrations were low (<0.6 mg O_2 L^{-1}) and hydrogen sulfide levels were high (Wurtsbaugh and Marcarelli 2004b). Additionally, when levels of salinity are below 5-6% the survival of brine shrimp may be reduced. Algal toxins that might be present in one or both bays could also contribute to mortality of juvenile brine shrimp. In addition to the laboratory and field experiments, I analyzed how the amount of screening on cages and their orientation in the water column influences brine shrimp survival within the cages.

Methods

Field Experiment–To determine survival and growth of brine shrimp in the Great Salt Lake, cages were constructed out of plastic bottles (Figure 1). The cages held approximately 2-L of water and had two 7x14 cm, 153µm mesh windows on either side. There were three cages per orientation (vertical or horizontal) for a total of 6 cages at each depth (0.2 m and 0.8 m). Cages were tied in milk crates and then placed into Farmington and Gilbert Bays at their respective depths on September 30, 2004. A total of 12 cages at each site were left in place for one week, when they were retrieved so growth and survival could be determined. In Farmington Bay the

cages were deployed ca. 1-km south of the causeway bridge where the water was 1-m deep. In Gilbert Bay, the cages were deployed on the north end of Antelope Island in water 1-m deep. The Gilbert Bay site likely periodically received plumes of water leaving Farmington Bay. At the end of the experiment nauplii were collected and preserved with a 3% formalin solution. In the lab samples were enumerated and nauplii were measured using a dissecting microscope to estimate growth.

Two-day old brine shrimp nauplii were used for the experiment. Nauplii were hatched at approx. 30°C in 3% NaCl. The nauplii that were to be used in Farmington Bay cages were transported to the lake in the 3% NaCl, while nauplii for Gilbert Bay cages were transported in 10% NaCl water to reduce stress affects when they were exposed to high salinity Gilbert Bay water. Nauplii were placed in cages at initial densities of 10 L^{-1} (20 $cage^{-1}$).

At the start of the experiment, temperature and dissolved oxygen were measured with a YSI model 58 DO/temperature meter at 1300 h in Farmington Bay and around 1600 at an offshore site in Gilbert Bay. We did not measure temperature and oxygen at the Gilbert Bay incubation site because of impending darkness. The water transparency was determined with a Secchi disk and depth was measured with a weighted line. Zooplankton densities were measured with a vertical tow from the bottom to the surface with a 0.5-m diameter 153-µm mesh zooplankton net.

At both the start and the end of the experiment we collected water with an 8-L horizontal Van Dorn sampler at both 0.2 m and 0.8 m to measure salinity, chlorophyll and hydrogen sulfide. For chlorophyll analysis 15–30 ml of water was filtered through GF/F filters, filters were wrapped in tinfoil and immediately placed on dry ice. Filters were subsequently extracted in 95% ethanol for 24 hr and chlorophyll *a* measured using a non-acidification technique with a Turner Model 10-AU fluorometer (Welschmeyer 1994). For hydrogen sulfide analysis water from the Van Dorn sampler was poured into BOD bottles using the triple over flow technique to minimize contamination by oxygen. Approximately 15 drops of zinc acetate was added to BOD bottles for preservation of hydrogen sulfide (APHA 1992). The concentrations of hydrogen sulfide were determined using the iodometric titration method (APHA 1992).

Laboratory Experiment–For the lab experiment, water from Farmington and Gilbert bays was collected on 30 September 2004 and the experiment started on 18 October. Water was collected with an 8-L horizontal Van Dorn sampler at the sites where the cages were located in Farmington Bay and Gilbert Bay, at depths of 0.2 m and 0.8 m. 2-day old brine shrimp nauplii were placed in 0.9-L glass bottles at initial densities of 10 L^{-1} and left in an environmental chamber at approximately 20°C with 12 light and 12 hrs dark for 1 week. Bottles were mixed and randomized at least twice daily. Three replicates were conducted for each depth and site, except for Farmington bay 0.8 m, where only two replicates were done and one of those replicates contained 0.54-L of lake water because not enough lake water was collected to achieve full replication.

Results

In the field experiment many extraneous aquatic invertebrates entered the experimental cages, including corixids and harpacticoid copepods in Farmington Bay and brine shrimp and brine fly larvae in Gilbert Bay. Cages with extraneous zooplankton were omitted from analysis because it was likely that the test nauplii could have escaped if other organisms entered (Table 1). Comparing the orientation of the field cages, a t–test assuming equal variances showed that there was no difference in nauplii survival between the two orientations ($p = 0.99$; Table 2).

Percent survival of brine shrimp nauplii in Farmington and Gilbert Bays was nearly the same at both depths (Figure 2). ANOVA analysis of percent survival revealed that there was no significant difference between the bays and depths ($F = 0.57$, $p = 0.65$; Table 3). The growth of

brine shrimp nauplii in Farmington and Gilbert bays was very similar (Figure 3). Nearly all of the biota that entered the cages in Farmington bay at 0.8 m were found dead, while almost all of the biota found in cages at 0.2 m were found alive (Figure 4).

The results from the laboratory experiment showed no nauplii survival in 0.2 m Gilbert Bay water and 0.8 m Farmington Bay water (Figure 5). The greatest survival occurred in 0.2 m Farmington Bay water. These differences in survival were significant (two-way ANOVA; $F = 5.22$, $P = 0.03$; Table 4). Post-hoc tukey analysis showed that survival of nauplii in 0.2 m Farmington Bay water was significantly different from both 0.2 m Gilbert Bay and 0.8 m Farmington Bay water, but not significantly different from 0.8 m Gilbert Bay water (Table 5).

In the laboratory experiment, nauplii approximately doubled in size during the 1-week trial. Nauplii in 0.2 m Farmington Bay water grew the most; however, because no nauplii survived in 0.2 m Gilbert Bay and 0.8 m Farmington Bay water, growth could not be determined for those groups (Figure 6). Because of the large error there was probably no significant difference in growth between 0.2 m Farmington Bay water and 0.8 m Gilbert Bay water.

The differences in brine shrimp survival may be due to the substantial difference in physical and chemical conditions between Farmington and Gilbert Bays. Salinities at the Gilbert Bay site were much higher (17%) than at the Farmington Bay site (3.4%; Figure 7). Salinities did not vary from the start to the end of the field experiment. At the start of the field experiment at an offshore site in Gilbert Bay, dissolved oxygen (2.7 mg L^{-1}) and temperature (18 ºC) remained constant throughout the water column (Figure 8). At the same time, water temperatures in Farmington Bay ranged from 16.8 ºC near the surface to 15.8 ºC at the bottom (Figure 9), indicating that even though the water column was shallow, there was still stratification. Dissolved oxygen in Farmington Bay ranged from 3.5 mg L^{-1}, near the surface to 0 mg L^{-1} below 0.6 m (Figure 9). This deep anoxic water had mean H_2S concentrations of 21.0 mg S L^{-1} (Figure 10). Hydrogen sulfide concentrations in Farmington Bay remained higher than concentrations in Gilbert Bay from the start to the end of the experiment (Figure 10).

Chlorophyll a concentrations were far higher in Farmington Bay than in Gilbert Bay (Figures 11, 12). In Farmington Bay the surface concentrations ranged from 80-120 µg L^{-1}, and they were even greater in the 0.8-m sample, particularly the one taken at the start of the experiment in the anoxic layer (516 µg L^{-1}; Figure 11). It is possible that this "chlorophyll" was actually bacteriochlorophyll. Chlorophyll *a* levels in Gilbert Bay were low, with the highest concentration (1.63 µg L^{-1}) at 0.2 m (Figure 12). Counts of phytoplankton done by the AWER 4510 class were consistent with the chlorophyll observations, and very few phytoplankton were found in the Gilbert Bay water. Green algae, diatoms and some cyanobacteria were found in the abundant plankton from Farmington Bay.

Discussion

No conclusive evidence was gained about survival and growth of brine shrimp in the Great Salt Lake with the field experiment. This was due to problems with cage construction and subsequent loss of data. If numerous aquatic invertebrates could get in the cages, then brine shrimp nauplii could surely have gotten out. However, the experiment did provide some interesting findings. Most of the aquatic invertebrates that entered the 0.8 m Farmington Bay cages were found dead (Figure 4). Since corixids are air breathers they likely were caught in the cages and then were not able to get to the surface to breathe. Harpacticoid copepod survival was poor in both the surface and deep cages in Farmington Bay. Of the five cages entered by the harpacticoids, only one had living harpacticoids at the end of the experiment. This could be due to anoxic conditions at the start of the experiment. It is not known how long the bottom water remained anoxic because we were unable to collect oxygen data at the end of the experiment, nor deploy an oxygen-recording sonde. In a laboratory experiment (Marcarelli et al. 2003) it was found that 100% mortality of brine shrimp nauplii occurred within 8 hr in

anoxic water. Another factor that can have detrimental effects on aquatic organisms is hydrogen sulfide in anoxic water. Watts (2001) found that sulfide in the range of 1-5 mg L^{-1} can be lethal to aquatic organisms. We found sulfide concentrations above this level both the start (21 mg L^{-1}) and end (8.0 mg L^{-1}; Figure 10) of the experiment in Farmington Bay. Another interesting observation is that some brine shrimp remained in the 0.8 m cages at the end of the experiment, despite the high hydrogen sulfide concentrations and low oxygen conditions (Figure 13). I assume that the brine shrimp found in the cages were nauplii that I put in the cages, rather than brine shrimp and nauplii that entered from the outside the cage, because zooplankton sampling by the AWER 4510 class on the first day of the experiment indicated that brine shrimp were absent from the Farmington Bay site (Figure 14). It is possible, however, that following sampling on September 30th brine shrimp drifted into Farmington Bay from Gilbert Bay.

In contrast to the field experiment, the laboratory experiment provided more conclusive evidence on how water quality influenced brine shrimp survival. For instance, 30% survival of brine shrimp in 0.2 m Farmington Bay water in contrast to the 0% survival in the 0.8 m water indicate that there was something in the deeper water that was lethal to the nauplii. Food levels, as measured by chlorophyll, were very high in both the surface and deep water of Farmington Bay, so the nauplii at either depth should not have starved to death, unless the phytoplankton were unpalatable. Because salinities in Farmington Bay are nearly equal at both depths, then we should have expected to see survival nearly equal.

It is not clear why no brine shrimp survived in the laboratory experiment when we used water from the surface waters of Gilbert Bay. Survival was also only 20% in the 0.8-m water from Gilbert Bay. It is possible that food levels in Gilbert Bay were too low and the nauplii starved to death. Overall survival in the experiment was low in all of the treatments, so it is possible that other factors caused mortality.

The most likely reason for poor survival of brine shrimp in the deep water from Farmington Bay was the lack of oxygen and high H_2S levels encountered in this water at the start of the experiment. Although we did not measure oxygen during the laboratory experiment, it is likely that the high sulfide concentrations in the deep water (21 mg L^{-1}) would have stripped any oxygen from the water. Low oxygen conditions and high hydrogen sulfide concentrations could surely have an effect on brine shrimp survival, as Watts et al. (2001) found that these conditions killed phytoplankton, zooplankton and fish when deep water in the Salton Sea was mixed into the water column.

In conclusion, this project provides evidence that hydrogen sulfide could be a factor that limits the abundance and distribution of brine shrimp in the Great Salt Lake. Additionally, experimental cages can be used to assess water quality, and with more planning time and a better design more conclusive results could have been acquired. More research is needed to understand how hydrogen sulfide or other factors in Farmington Bay affect brine shrimp.

References

APHA. 1992. Standard methods for examination of water and wastewater. Eighteenth edition. American Public Health Association, Washington D.C.

Nicholson B and Marcarelli AM. 2004. The paradox of a Great Salt Lake. Southwest Hydrology 3: 24-25.

Marcarelli A., W.A. Wurtsbaugh, B. Albrecht, E. Archer, J. Basset, M. Beckstrand, M. Hadley, J. Kling, O. Lester, P. MacKinnon, T. Ting. 2003. Continuing studies of water quality in Farmington Bay and the Great Salt Lake. Aquatic Ecology Practicum Class Project 2002. College of Natural Resources, Utah Sate University. 91p,

Watts J.M., B.K. Swan, M.A. Tiffany and S.H. Hurlbert, 2001. Thermal, mixing, and oxygen regimes in the Salton Sea, California, 1997-1999. Hydrobiologia 466: 159-176.

Welschmeyer N. A. 1994. Fluorometric analysis of chlorophyll *a* in the presence of chlorophyll *b* and pheopigments. Limnology and Oceanography 39: 1985-1992.

Wurtsbaugh, W.A., A. Marcarelli, D Gross, J Moore, C Christison, S Archer, and S Bates. 2002. Comparative analysis of pollution in Farmington Bay and the Great Salt Lake, Utah. Aquatic Ecology Laboratory Class Project 2001. College of Natural Resources, Utah Sate University. 21p.

Wurtsbaugh, WA and AM Marcarelli. 2004a. Phytoplankton and zooplankton in Farmington Bay and the Great Salt Lake, Utah (2003). Report to the Central Davis Sewage Improvement District, Kaysville, UT. 57 p.

Wurtsbaugh, WA and AM Marcarelli. 2004b. Hydrogen sulfide in Farmington Bay and the Great Salt Lake: a potential odor-causing agent. Report to the Central Davis Sewage Improvement District, Kaysville, UT. 30 p.

Tables

Table 1. Data from the field experiment showing the number of brine shrimp alive or found dead at the end of a 1-week trial in Farmington or Gilbert Bays. Highlighted rows were omitted from the analysis because extraneous zooplankton got into the cages.

Bay	Orientation	Depth of cage (m)	rep. #	Nauplii # alive	Nauplii # dead	% survival	Growth (mm)	Comments
Farmington	Horizontal	0.2	2	0		0		5 CORIXID (ALIVE), 22 HARPACT.(DEAD)
Farmington	Horizontal	0.2	1	0		0		12 CORXID (ALIVE), FEW HARPACT.(DEAD)
Farmington	Horizontal	0.2	3	0		0		ALIVE CORIXD, HARPACT ALIVE AND DEAD
Farmington	Vertical	0.2	1	11		0.55	0.9	
Farmington	Vertical	0.2	3	0	5	0		
Farmington	Vertical	0.2	2	7	1	0.35	0.9	HARPACT 3 ALIVE AND 3 DEAD
Farmington	Horizontal	0.8	2	0		0		
Farmington	Horizontal	0.8	3	1		0.05	1.4	4 CORIXD (DEAD), HARPACT (DEAD).
Farmington	Horizontal	0.8	1	0		0		HARPACT (DEAD)
Farmington	Vertical	0.8	3	0		0		HARPACT (DEAD)
Farmington	Vertical	0.8	2	10	3	0.5	1.4	HARPACT (DEAD)
Farmington	Vertical	0.8	1	1		0.05	0.9	Many corixid dead and many Harpact alive
Gilbert	Horizontal	0.2	2	120	17	6	3.5	1brine fly larvae
Gilbert	Horizontal	0.2	1	6		0.3	4.1	hole in plastic
Gilbert	Horizontal	0.2	3	1		0.05	0.9	1brine fly larvae
Gilbert	Vertical	0.2	2	1		0.05	0.6	
Gilbert	Vertical	0.2	3	10		0.5	1.4	
Gilbert	Vertical	0.2	1	27		1.35	6.6	
Gilbert	Horizontal	0.8	1	107	1	5.35	6.9	lid off, 1brine fly larvae
Gilbert	Horizontal	0.8	2	20		1	1.4	
Gilbert	Horizontal	0.8	3	0		0		
Gilbert	Vertical	0.8	2	1		0.05	0.8	
Gilbert	Vertical	0.8	1	0		0		
Gilbert	Vertical	0.8	3	3		0.15	1.8	

Table 2. Two-Sample T-test assuming Equal Variances comparing the percent survival of brine shrimp nauplii between cages deployed in two different orientations.

	Horizontal	*Vertical*
Mean	0.33	0.33
Variance	0.33	0.22
Observations	3	8
Pooled Variance	0.24	
Hypothesized Mean Difference	0	
Df	9	
t Stat	0.006	
P(T<=t) one-tail	0.497	
t Critical one-tail	1.833	
P(T<=t) two-tail	0.995	
t Critical two-tail	2.262	

Table 3. Two-way ANOVA for field data with bay and depth as factors and survival as the response variable.

Class	Levels	Values
bay	2	FB GB
depth	2	0.2 0.8

Number of observations 11

Dependent Variable: survival

Source	DF	Sum of Squares	Mean Square	**F Value**	**Pr > F**
Model	3	0.431	0.143	**0.57**	**0.6512**
Error	7	1.759	0.251		
Corrected Total	10	2.191			

R-Square	Coeff Var	Root MSE	survival Mean
0.196885	151.1113	0.501415	0.331818

Source	DF	Type III SS	Mean Square	**F Value**	Pr > F
Bay	1	0.17606694	0.176	**0.70**	0.4303
Depth	1	0.21967350	0.219	**0.87**	0.3810
Bay*Depth	1	0.00688661	0.006	**0.03**	0.8732

Table 4. Two-way ANOVA for lab data with depth and bay as factors and survival as the response variable.

Class	Levels	Values
bay	2	FB GB
depth	2	0.2 0.8

Number of observations 11

Dependent Variable: survival

Source	DF	Sum of Squares	Mean Square	F Value	Pr > F
Model	3	0.26102424	0.08700808	**5.22**	**0.0332**
Error	7	0.11666667	0.01666667		
Corrected Total	10	0.37769091			

R-Square	Coeff Var	Root MSE	survival Mean
0.691105	85.54782	0.129099	0.150909

Source	DF	Type III SS	Mean Square	F Value	Pr > F
bay	1	0.02322963	0.02322963	1.39	0.2763
depth	1	0.02322963	0.02322963	1.39	0.2763
bay*depth	1	0.20411852	0.20411852	12.25	0.0100

Table 5. Post Hoc TUKEY analysis for survival of nauplii in the lab experiment.

Adjustment for Multiple Comparisons: Tukey-Kramer

bay	depth	survival LSMEAN	LSMEAN Number
FB	0.2	0.37000000	1
FB	0.8	-0.00000000	2
GB	0.2	0.00000000	3
GB	0.8	0.18333333	4

Least Squares Means for effect bay*depth
Pr > |t| for H0: LSMean(i)=LSMean(j)
Dependent Variable: survival

i/j	1	2	3	4
1		0.0624	0.0387	0.3597
2	0.0624		1.0000	0.4576
3	0.0387	1.0000		0.3731
4	0.3597	0.4576	0.3731	

Figures

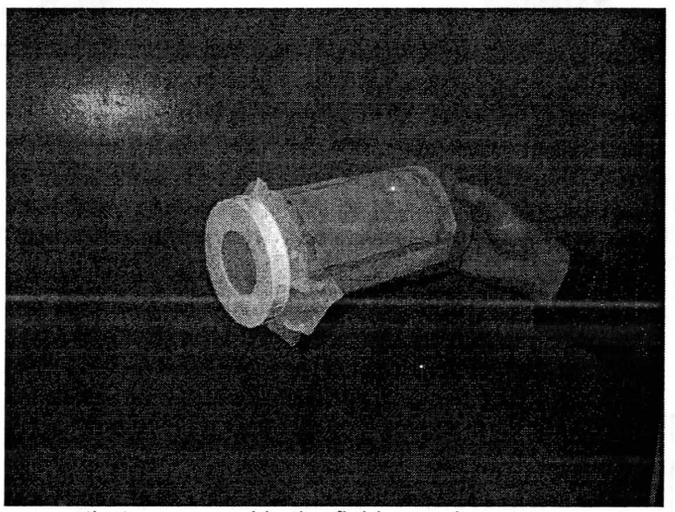

Figure 1. Picture of the cages that were used in the field experiment.

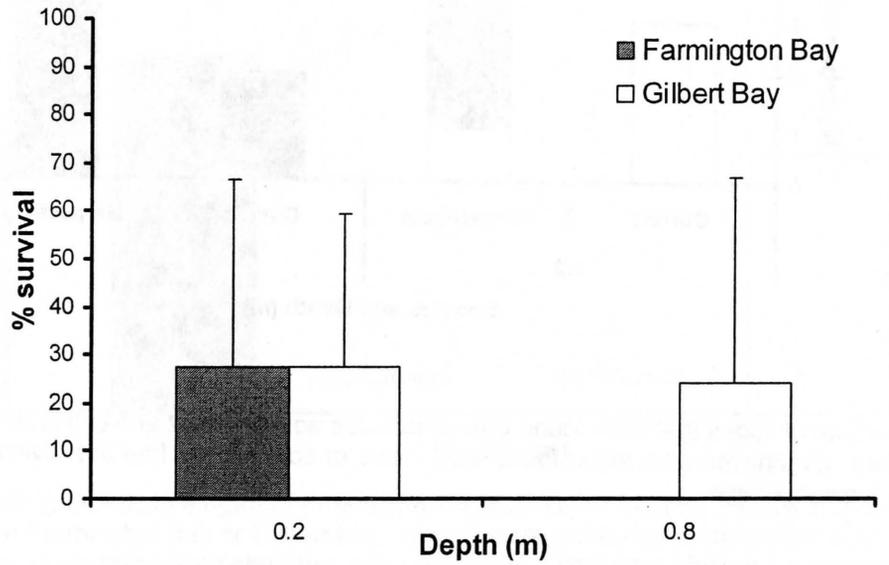

Figure 2. Percent survival of *Artemia franciscana* nauplii held for 1-week in meshed cages in Farmington Bay and Gilbert Bay of the Great Salt Lake. The cages were incubated at two depths. The highlighted data in Table 1 were omitted. Error bars show ± 1 standard deviation.

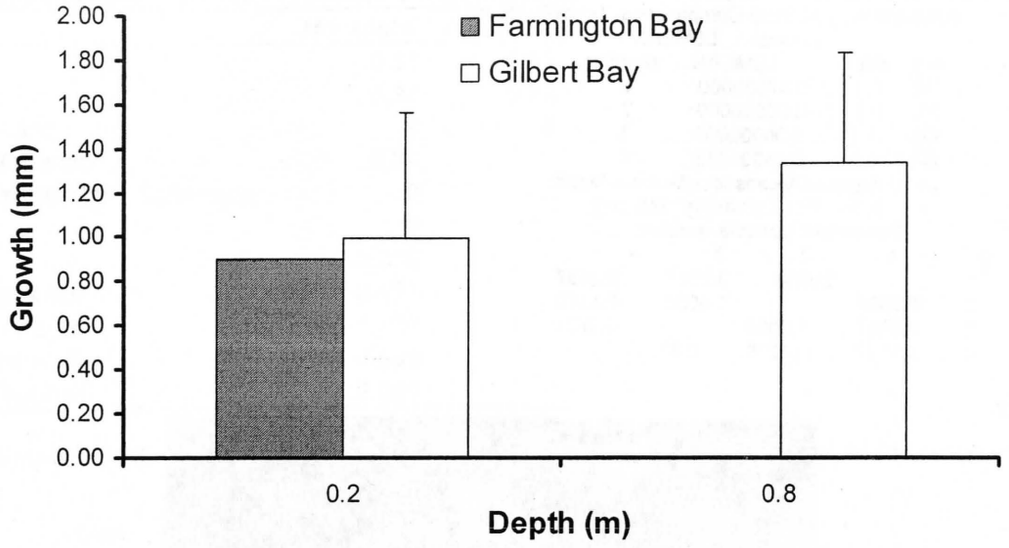

Figure 3. Growth (increase in length) of Artemia *franciscana* nauplii held for 1-week in meshed cages in Farmington Bay and Gilbert Bay of the Great Salt Lake. The cages were incubated at two depths. The highlighted data in Table 1 were omitted. Error bars show ± 1 standard deviation.

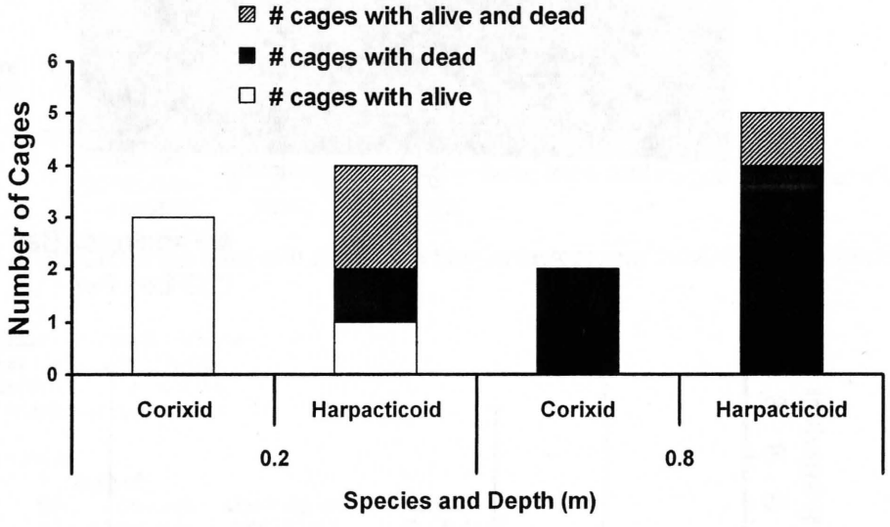

Figure 4. Number of cages that were found with extraneous aquatic life at end of incubation period in Farmington Bay. Organisms were either found alive, dead, or some cages had both living and dead organisms in the same cage.

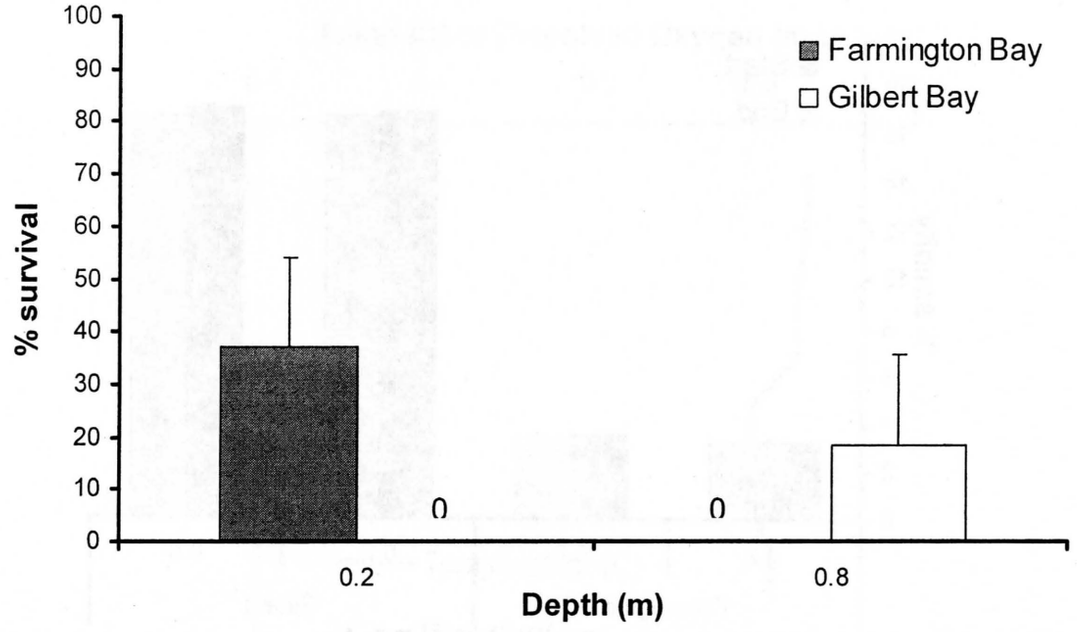

Figure 5. Percent survival of *Artemia franciscana* nauplii in lab study that were held for 1-week in bottles in lake water from either Farmington or Gilbert Bay. Nauplii were incubated in lake water from two depths and were held in an environmental chamber in the laboratory. Error bars show ± 1 standard deviation.

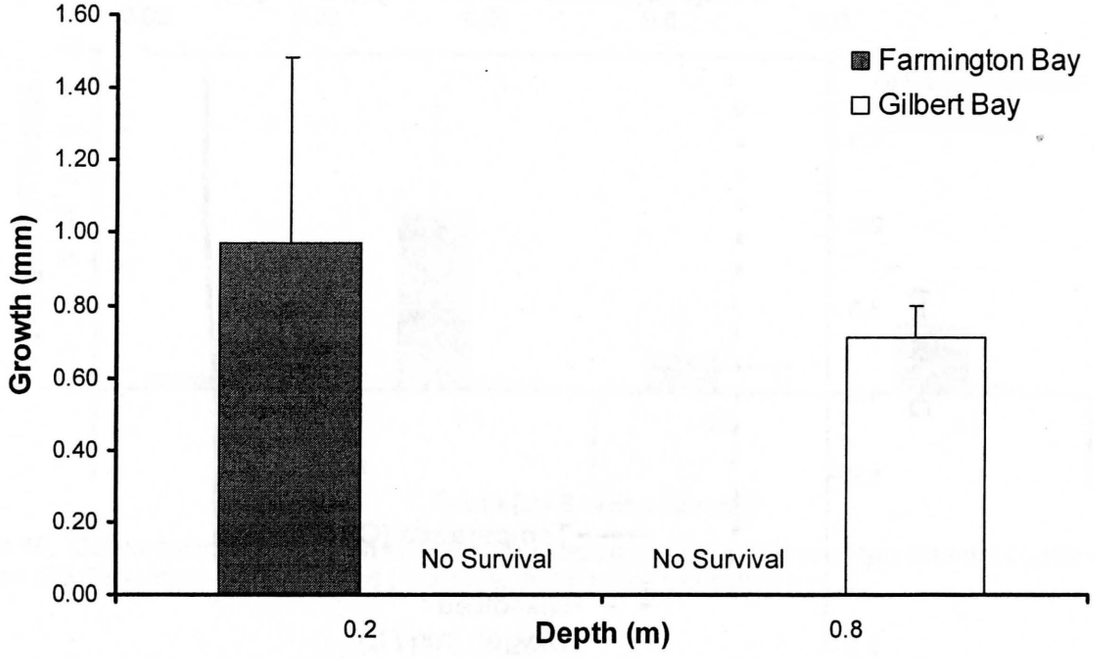

Figure 6. Growth (increase in length) of Artemia *franciscana* nauplii held for 1-week in bottles of lake water from either Farmington Bay or Gilbert Bay. Nauplii were incubated in lake water from two depths and were held in an environmental chamber in the laboratory. Error bars show ± 1 standard deviation.

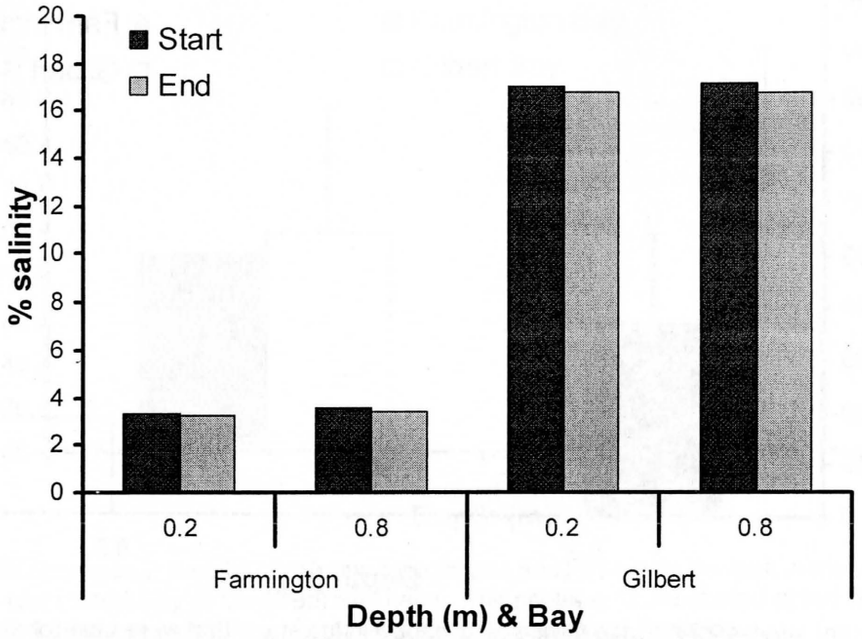

Figure 7. Percent salinities measured in Farmington and Gilbert Bays at two depths at the start (30 September 2004) and end (7 October 2004) of the incubation period.

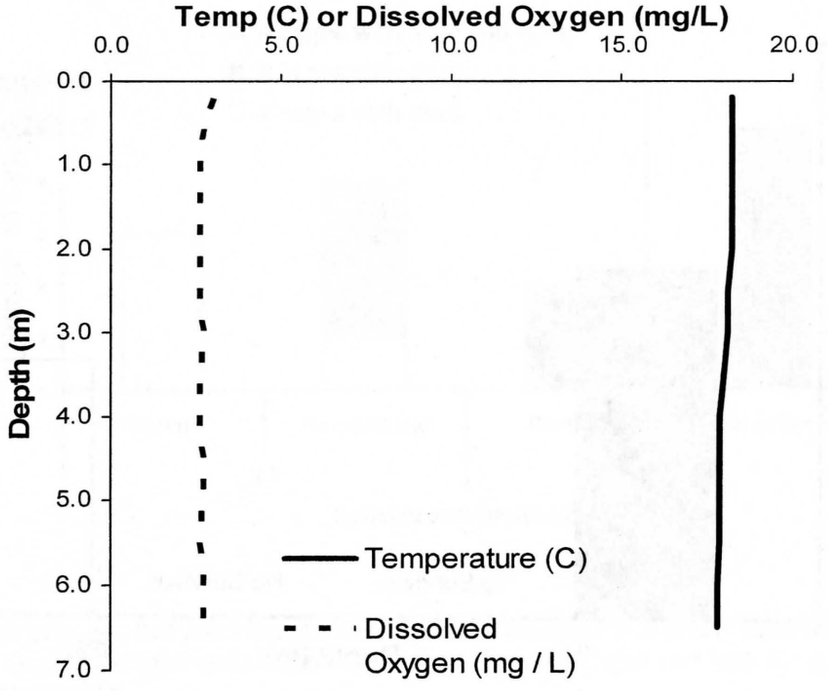

Figure 8. Temperature and dissolved oxygen profiles in Gilbert Bay. Profiles were collected at the start (30 September 2004) of the incubation period.

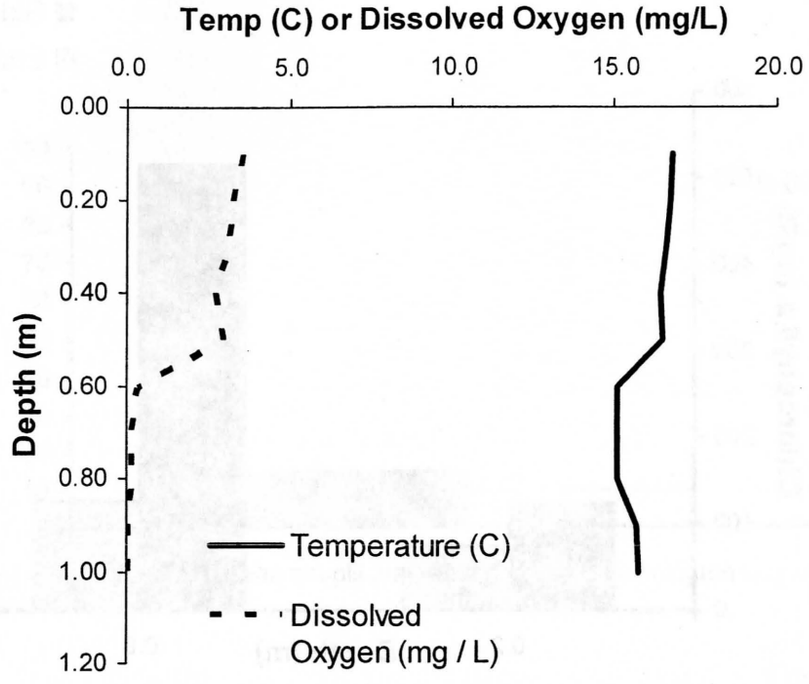

Figure 9. Temperature and dissolved oxygen profiles in Farmington Bay. Profiles were collected at the start (30 September 2004) of the incubation period.

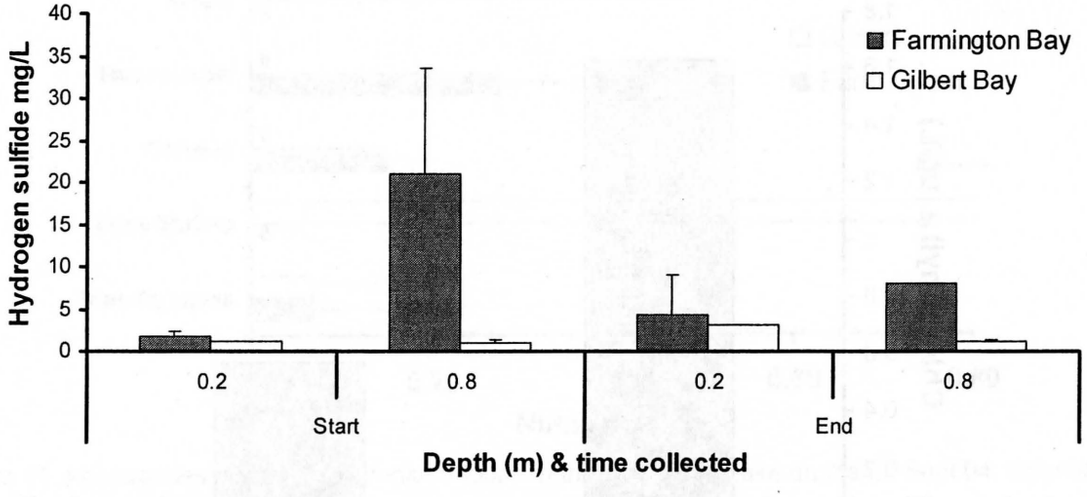

Figure 10. Concentration of hydrogen sulfide in Farmington and Gilbert Bays at two different depths at the start (30 September 2004) and end (7 October 2004) of the incubation period.

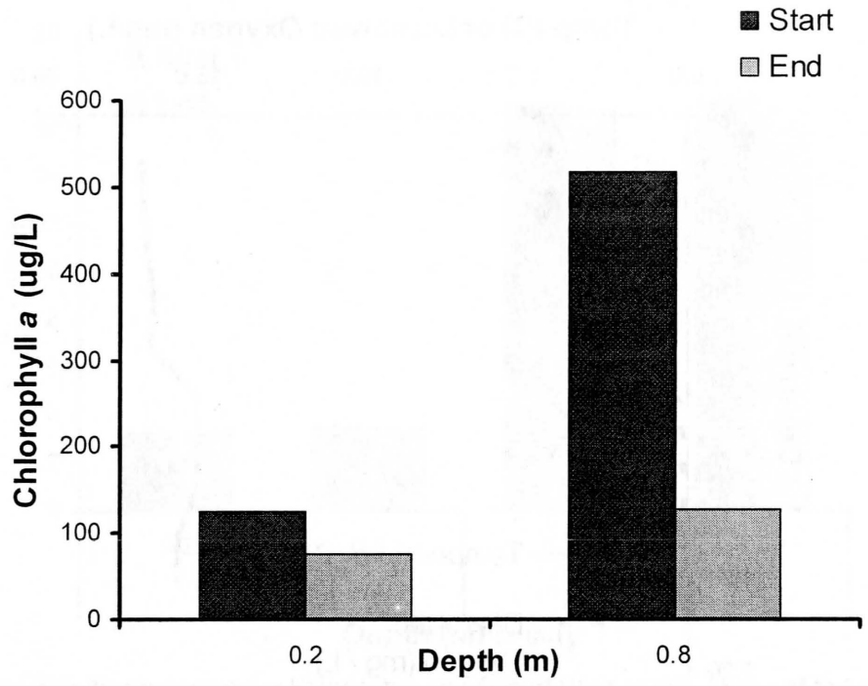

Figure 11. Chlorophyll *a* concentrations in Farmington Bay at two different depths. Chlorophyll *a* was measured at the start (30 September 2004) and end (7 October 2004) on the incubation period.

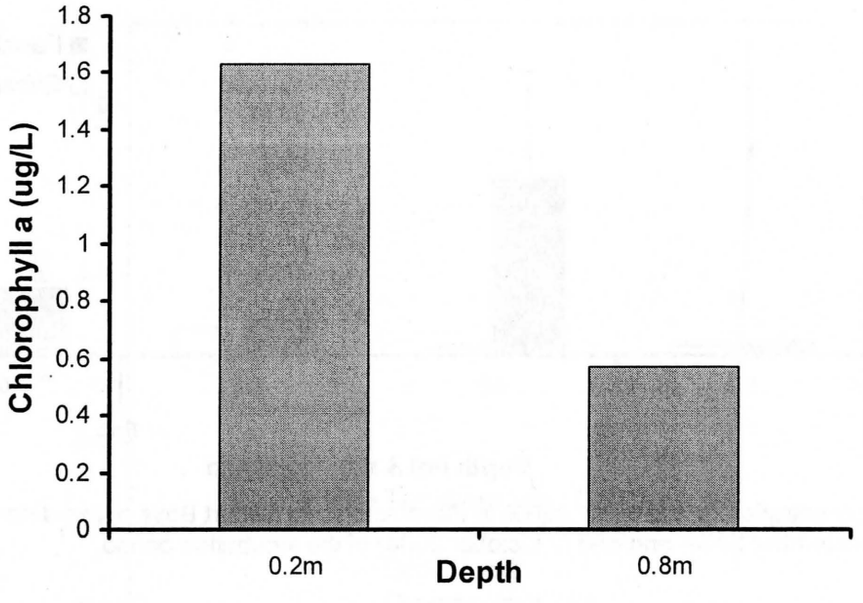

Figure 12. Chlorophyll *a* concentrations in Gilbert Bay at two different depths. Chlorophyll *a* was measured at the start (30 September 2004) of the incubation period. Note the different the different scale of the y-axis as compared to Figure 6.

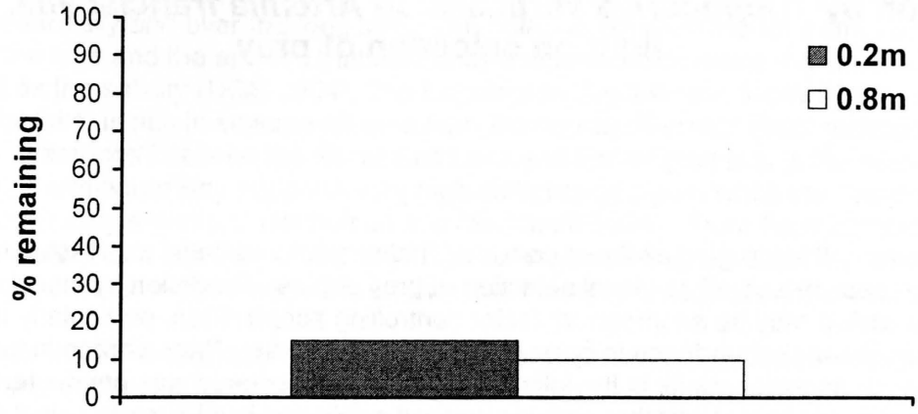

Figure 13. Percent of *Artemia franciscana* nauplii that remained in the Farmington Bay cages after the 1 week incubation period.

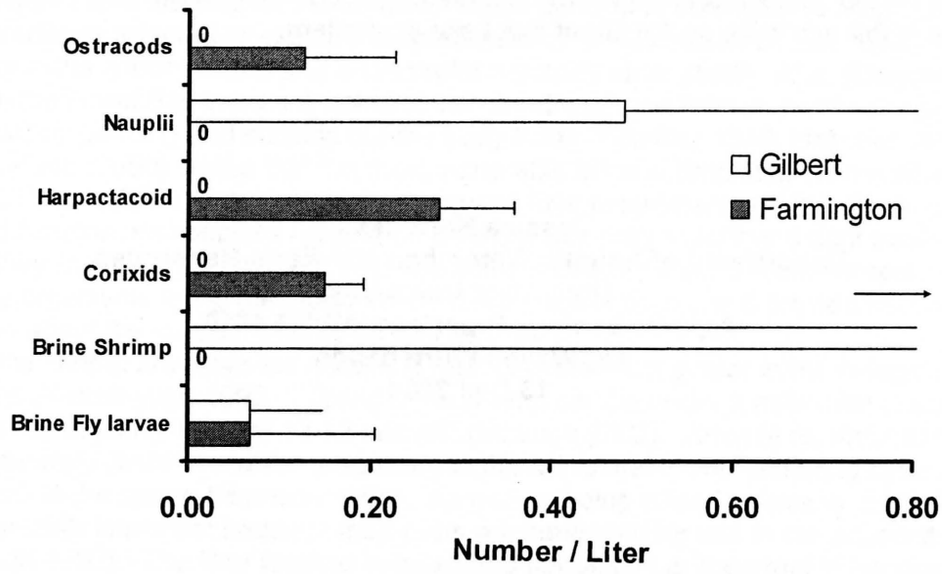

Figure 14. Abundances of macrozooplankton found in the Great Salt Lake on the 30 Sept 04. Error bars show ± 1 standard deviation.

Chapter 6

Predation by *Trichocorixa verticalis* on *Artemia franciscana*: effect of light on selection of prey

Summary

In this study the foraging ability of corixids (*Trichocorixa verticalis*) was tested to determine if mechanoreception as well as visual selection of prey occurs. Predation by insect species such as the corixid may be an important factor controlling zooplankton, particularly brine shrimp (*Artemia franciscana*) abundance in parts of the Great Salt Lake. Brine shrimp in turn can affect conditions such as water clarity in the lake. A laboratory experiment was conducted with light and dark treatments to test whether corixids depend solely on visual cues to select for prey or if tactile sensation can be used. The inability of corixids to feed on small organisms (brine shrimp nauplii) in the dark treatment was found to be moderately significant, thus potentially having an effect on brine shrimp size structure. Further research, including field experiments, is needed to determine if corixid tactile response to prey is more than moderately significant and what the implications of this would be on the Great Salt Lake ecosystem.

Jessica Horrocks
Department of Aquatic Watershed and Earth Resources
Utah State University

Aquatic Ecology Practicum AWER 4510
Dr. Wayne Wurtsbaugh
13 Dec 2004

Introduction

The Great Salt Lake is a terminal fishless lake located in northern Utah. The Utah Geological Survey reports it to be the 33rd largest lake in the world, and the largest fresh or saltwater lake in the United States after the Great Lakes. Size and depth of the lake, however, vary both seasonally and over the long term. The balance between the total amount of water that enters the lake and the amount that leaves by evaporation changes the dimensions of the lake as well as the salinity (UGS 2004). The Farmington Bay estuary, located in the southeast portion of the lake, is rich in sewage effluent from the nearby Wasatch Front metropolitan area. The biggest difference between the Great Salt Lake and Farmington Bay is the amount of productivity. Farmington Bay supports very high densities of algae, while the Great Salt Lake has much lower concentrations (Nicholson and Marcarelli 2004). From Farmington Bay (low salinity) the water flows into the Gilbert Bay (high salinity) of the Great Salt Lake through a very narrow passage under a causeway. Gilbert Bay and Farmington Bay are the areas of interest in my study.

Insects in both freshwater and saline waters are a common part of the littoral community, with the most prominent members being Hemiptera (Corixidae, Notonectidae; Hammer 1986). Corixids are among the most abundant of all aquatic macrofauna and occur in saline lakes on all continents except Antarctica (Hammer 1986). The Hemipteran found in the Great Salt Lake is not in the Family Notonectidae but the Family Corixidae, species name *Trichorixia verticalis*. Notonectids prey on planktonic crustaceans, rotifers and a variety of littoral organisms, and terrestrial organisms trapped on the surface. Some species of corixids and notonectids are cruising predators adapted to exploit the open water of fishless water bodies. They feed on active prey in the water column and often prefer relatively large zooplankton (Dieguez 2003). Corixids in the Great Salt Lake are nektonic (air-breathing) and feed on brine shrimp (*Artemia franciscana*) by piercing and sucking out the body fluids (Scudder 1976; Hammer 1986; Hadley 2002). Mellison (2000) stated that the most vulnerable brine shrimp stage is the small nauplii. Dieguez & Gilbert (2003) conducted laboratory and field experiments on the efficiency of the notonectid *Buenoa macrotibialis* feeding on different size prey in light and dark treatments. In contrast, they found that *B. macrotibialis*, like many predatory insects, could detect and feed on larger prey organisms using mechanoreceptors that detect waves and provide accurate information about the location of prey as well as vision.

Brine shrimp are often the dominate macrozooplankton grazer in the pelagic region of saline lakes (Wurtsbaugh 1992). Young brine shrimp can be under a millimeter (Hadley 2002) and adults can reach lengths of 10-12 mm (Wurtsbaugh 2002). *Artemia* reproduction can occur parthenogenically (without males) or sexually in the fall to make over-wintering cysts from which nauplii hatch in the spring (Harrison 1998). As well as being a food source for corixids, *Artemia* are economically important because their cysts are harvested for use in the aquaculture industry (Wurtsbaugh 1992). The filter feeding brine shrimp population is important in keeping the waters of the Great Salt Lake clean through algae consumption. They are also a key food source for millions of migrating birds (UGS 2004).

The purpose of my study was to test the hypothesis that the corixid can use tactile as well as visual cues when foraging for prey. The Great Salt Lake is hypereutrophic and therefore turbid; a characteristic to acquire prey other than vision, like mechanoreception would greatly enhance foraging ability. I predict to see a statistically significant positive correlation between prey size and those detected and fed upon in the dark by corixids.

Methods

Field Collections–Zooplankton samples were collected by the AWER 4510 class from Farmington and Gilbert Bay on 30 September 2004 to determine organism densities. A 50-cm diameter 153µm mesh zooplankton net was used. The haul depth was 0.9 m and 1.0 m in Farmington Bay and 6.2 m in Gilbert Bay. All samples were preserved in formalin and taken back to Utah State University to be counted. The zooplankton net hauls resulted in very few corixids and no brine shrimp for Farmington Bay and no corixids and many brine shrimp for Gilbert Bay (Table 1 and Figure 2). On the Farmington Bay side of the causeway there was a concentration of corixids immediately next to shore. This is where I used a hand net to collect corixids for use in my laboratory experiment. These live corixids were placed in plastic containers and transported back to the Utah State Limnology Laboratory.

Experimental Design–To test whether light availability impacted the ability of the corixids to select different sized *Artemia* I set up an experiment with a light treatment, a dark treatment and a control, each with three replicates. Each replicate consisted of a 12-L bucket filled with 5 L of 6% salt solution and 5% filtered Gilbert Bay water to provide algae as sustenance. Dark treatment buckets were blacked out using aluminum foil and the light intensities in the light buckets were measured with a LiCor radiometer. To keep each bucket properly oxygenated aeration devices were attached and running prior to the experiment, but not during the experiment. The containers were held at 20°C in an incubation room.

Artemia cysts were hatched in the incubation room and allowed to grow to a determined size. I chose to work with 1-mm or slightly smaller nauplii and 5-8 mm adult brine shrimp based on the experiments of Dieguez (2003) that suggested that notonectids rely on mechanoreception to capture prey larger than 1-mm. Fifty nauplii and thirty adult *Artemia* of the appropriate sizes were counted and added to each treatment bucket. *Artemia* densities in the experiment were consequently higher than those found in the Great Salt Lake (Table 1). I didn't want the corixids to be limited by prey available; also, numbers needed to be high to account for the natural mortality of prey (Wurtsbaugh pers. comm.). One corixid per bucket (0.2 L^{-1}) was added to the light and dark treatments for a corixid density almost equal to those found in Farmington Bay (0.18 L^{-1}) in late September (Table 1 and 2). The control treatments were incubated in the light with no predator in order to control for natural mortality of prey. After an exposure time of twelve hours the corixids were removed, the bucket water filtered through 70µm mesh, and the remaining brine shrimp were counted.

Calculations–After counting, I calculated the % mortality of adults and nauplii as well as the corixid clearance rate in each replicate. The equation for clearance rate is as follows (Hadley 2002):

$k = (\ln Pi - \ln Pf) / xt$

Where: Pi = final # of prey in controls (mean)
Pf = final # of prey in predator treatments
x = # of predators per liter
t = days

Clearance rate estimates the number of liters of water cleared of prey by a predator per day, and my calculation method also corrects for mortalities in control treatments.
Next I ran a two-way ANOVA using SAS V. 8e (PROC GLM) with size and light treatment as factors and % mortality as the response variable to determine if my results corresponded with my hypothesis in a statistically significant way (Appendix 1).

Results

Overall, in the light treatments the corixids selected more nauplii than adults, and in the dark treatments the corixids selected more adults than nauplii (Figure 2). The overall experimental results are moderately significant with a P value of 0.07 (F = 2.92, Appendix 1). The effect of the size of brine shrimp on their loss to predators was significant at p = 0.04. Figure 2 indicates that adult mortalities are highest in the dark and nauplii mortalities are highest in the light and lowest in the dark. Mean mortalities for light and dark are opposite for juveniles and adults, further supporting my hypothesis.

In the light, corixids cleared 0.6-L of water of adult brine shrimp. In the dark, presumably using tactile senses, they cleared an average of 3.5 L of adult brine shrimp. Calculation of corixid clearance rates feeding on brine shrimp nauplii were confounded by high mortality rates in the control treatments. Estimated mean clearance rates were 1.7 and –1.1 L predator^{-1} day^{-1} in the light and dark treatments, respectively (Table 2).

Discussion

My experimental results indicate that *Trichocorixa verticalis* may use mechanoreception when foraging for prey. I obtained higher percent mortalities for nauplii in the light than in the dark, suggesting that corixids cannot detect the small nauplii prey in the dark (Figure 2). In order to prove that corixids do indeed rely on mechanoreception further research and field experiments need to be conducted. My experiment was limited to few replicates, one of which failed (dark treatment A), therefore introducing a large probability of error. Also, only one type of prey at densities not encountered by corixids in Farmington Bay or Gilbert Bay was used (Table 1 and Table 2). An additional problem was that mortalities of nauplii in the control treatments were high, making it difficult to assess the impacts of the corixid predators.

Corixids can have a direct effect on the brine shrimp population (Figure 3; Marcarelli and Wurtsbaugh 2004). It has been observed in other field studies that shortly after corixid numbers peak, the brine shrimp population falls. This top-down control of upper levels of the food chain on the secondary producers (*Artemia*) and in turn on primary producers is defined as a trophic cascade (Dodds 2002). To better understand the trophic effects of corixids I compared their clearance rates between the different size classes and the different experimental treatments (Table 2). The clearance rates quantify how many liters of water a predator can clear of prey of each size class per day (Hadley 2002). The values suggest corixids have a significant impact on the waters they inhabit. At the densities of corixids encountered in Farmington Bay in September (0.18 L^{-1}), these predators could clear approximately 10% of the water column per day in the light, and 60% at night, giving an overall 24-hour day rate of approximately 35% on adult brine shrimp. This predation rate might be higher than can be sustained by brine shrimp populations (Hadley 2002). During years when corixids invaded the pelagic region of the Great Salt Lake due to lower salinity levels the brine shrimp population decreased and the conditions of the lake changed drastically. *Artemia* biomass dropped from 720 to 2 mg m^{-3} and consequently there was a 10-fold decrease in community filtration rate, in turn contributing to a 20-fold increase in chlorophyll *a* concentration, and a 4-fold decrease in water clarity (Wurtsbaugh 1992). The physical change of lower salinity lead to an increase in corixid populations, causing a trophic cascade that ultimately affected the water quality of Great Salt Lake. One should note, however, that alternative hypotheses for the decline of brine shrimp during the low-salinity period have been forwarded (Stephens 1990).

It is important to understand how corixids select for prey because invertebrate predation may be important in structuring the food web in the Great Salt Lake (Wurtsbaugh 1992), and should be considered when making management decisions. Managers can take advantage of trophic cascades by using biomanipulation to improve water quality of lakes (Dodds 2002). The

method of feeding for corixids could help in understanding the predator/prey interaction between corixids and brine shrimp and subsequent lake effects including water quality. With small prey being vulnerable in the light (Mellison 2000) and undetectable by predators in the dark (Dieguez 2003) the size structure of *Artemia* may vary depending on light and turbidity in the Great Salt Lake. Further research, including field experiments, is needed to determine if corixid tactile response to prey is more than moderately significant and what the implications of this would be on the Great Salt Lake.

References

Dieguez, M. and J. Gilbert. 2003. Predation by *Buenoa macrotibialis* (Insecta, Hemiptera) on zooplankton: effect of light on selection and consumption of prey. Journal of Plankton Research, 25: 759-769.

Dodds, W.K. 2002. Freshwater Ecology: Concepts and Environmental Applications. San Diego: Academic Press.

Hadley, M. The impact of predation by corixids on brine shrimp in Farmington Bay of the Great Salt Lake, Utah. *In* A. M. Marcarelli, W.A. Wurtsbaugh, B. Albrecht, E. Archer, J. Bassett, M. Beckstrand, M. Hadley, J. Kling, O. Lester, P. MacKinnon and T-h Ting. 2003. Continuing studies of water quality in Farmington Bay and the Great Salt Lake, Utah. Aquatic Ecology Practicum Class Project 2002. College of Natural Resources, Utah State University. 91 p.

Hammer, T. 1986. Saline Lake Ecosystems of the World. Dordrecht: DR W. Junk Publishers.

Harrison, T. 1998. General Ecology Class Project. Westminster College. Retrieved December 5, 2004 from the World Wide Web: http://people.westminstercollege.edu/faculty/tharrison/gslfood/studentpages/brine.html

Marcarelli, A. and W. A. Wurtsbaugh. 2004. Predation by an invertebrate predator in Farmington Bay, Great Salt Lake, Utah. Utah State University Runoff Conference Poster, Logan, UT.

Mellison, S.C. 2000. Functional response of a water boatman (*Trichocorixa verticalis*) and environmental conditions that affect its distribution in the Great Salt Lake, Utah, USA. Thesis. Utah State University, 2000.

Nicholson, B. and A.M. Marcarelli. 2004. The paradox of a Great Salt Lake. Southwest Hydrology 3 (4): 24-25.

Scudder, G.G.E. 1976. Water-boatmen of saline waters (Hemiptera: Corixidae). North-Holland, pp 263-289.

Stephens, D.W. 1990. Changes in the lake levels, salinity and the biological community of the Great Salt Lake, Utah, USA (1947-1987). Hydrobiologia 197: 130-146.

Utah Geological Survey. 2004. Commonly Asked Question about Utah's Great Salt Lake and Lake Bonneville. State of Utah. Retrieved December 5, 2004 from the World Wide Web: http://www.ugs.state.ut.us/index.htm

Wurtsbaugh. W. A. 1992. Food-web modification by an invertebrate predator in the Great Salt Lake (USA). Oecologia, 89: 168-175.

Figures

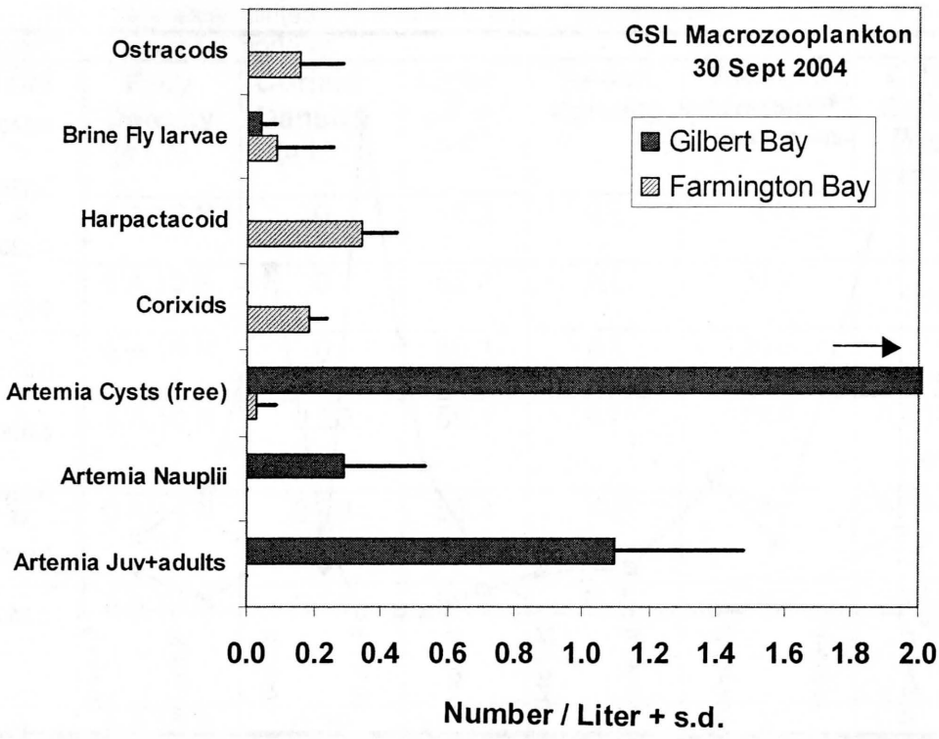

Figure 1. Densities of brine shrimp in Gilbert and Farmington Bays on 30 September 2004. Collections and counts were done by the AWER 4510 class.

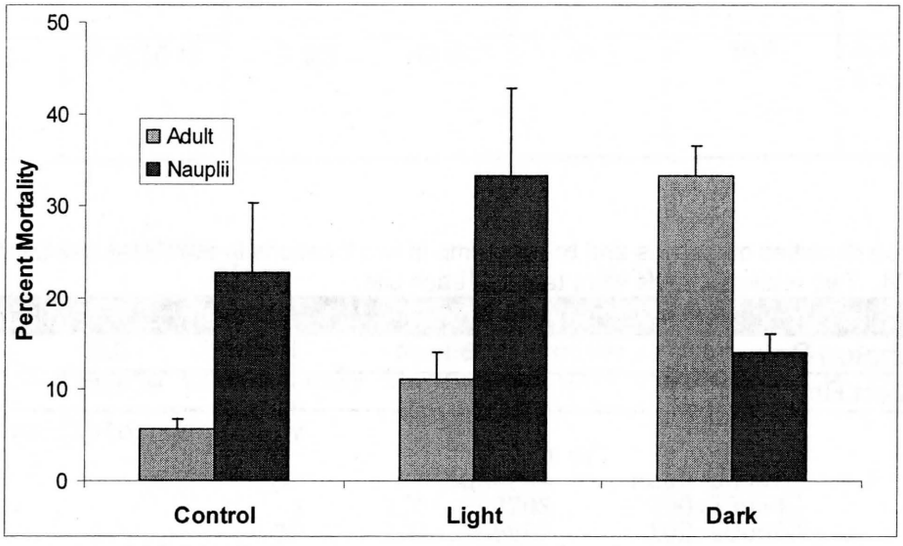

Figure 2. Results of corixid detection and capture of *Artemia franciscana* prey in light and dark treatments. Percent mortality was calculated from the average of three replicates for light and control treatments, and the average of two replicates for the dark treatment. Note the opposing results for the light and dark treatments. Error bars indicate one standard error. In the two-way ANOVA, treatment (control, light, dark) was not significant ($p = 0.40$), stage (adult, nauplii) was significant at $p = 0.04$, and there was a nearly significant interaction between these two factors ($p = 0.07$).

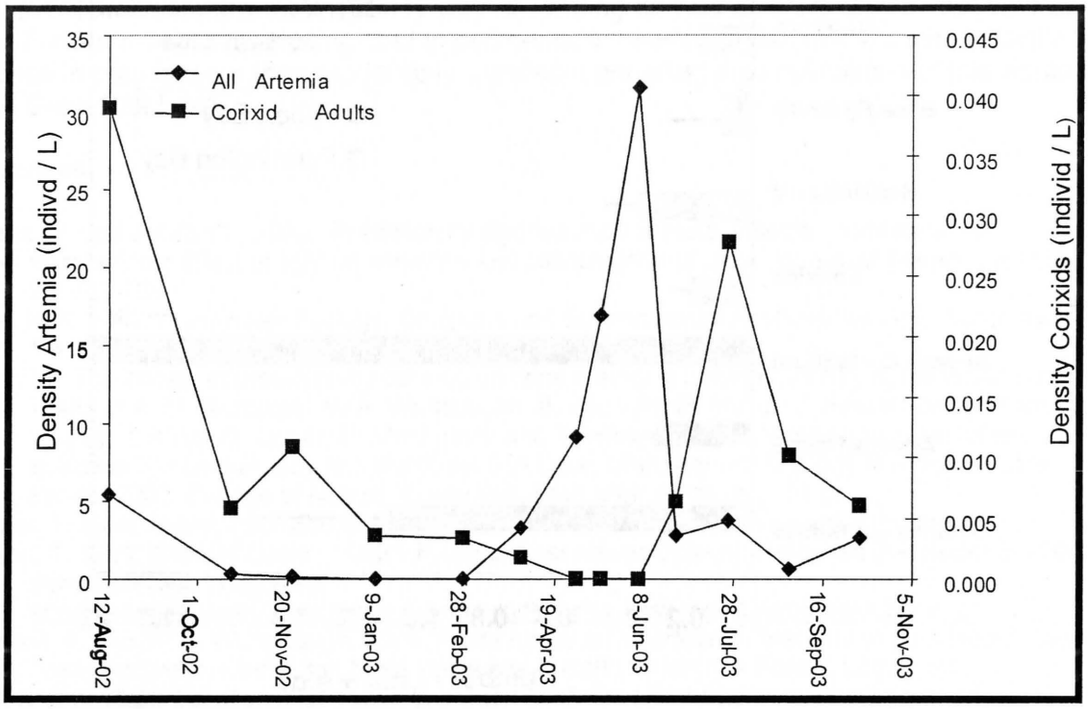

Figure 3. Densities of *Artemia franciscana* and corixids in Farmington Bay in 2002 and 2003. Note the rapid decline in *Artemia* when there is a peak in corixid density. Data from Marcarelli and Wurtsbaugh (2004).

Tables

Table 1. Mean densities of corixids and brine shrimp in two locations in the Great Salt Lake on 30-September-04. Two replicate hauls were taken at each site.

Site	Density of Corixids (# L^{-1})	Density of Artemia (# L^{-1})
Farmington Bay	0.18	0.00
Gilbert Bay	0.00	1.83

Table 2. Information on the laboratory experiment comparing light treatments (C control (no predator), L light, D dark) to size selection of prey (A adult, N nauplii) by adult corixids. All experiments contained 5%, 70-µm filtered Gilbert Bay water diluted to 6% salinity, and an exposure time of 12 hours. X's indicate a failed experiment due to corixid mortality.

Treatment	Trial	Prey Density (# L^{-1})	Corixid Density (# L^{-1})	Light (µE m^{-2} sec^{-1})	%Adult Mortality	%Nauplii Mortality	Clearance Rate (Liters cleared $predator^{-1}$ d^{-1})
C	A	6 A;10 N	0	56.1	6.7	8.0	0
C	B	6 A;10 N	0	56.1	6.7	34.0	0
C	C	6 A;10 N	0	56.1	3.3	26.0	0
L	A	6 A;10 N	0.20	56.1	6.7	52.0	A 0.12;N 4.77
L	B	6 A;10 N	0.20	56.1	10.0	28.0	A 0.48;N 0.72
L	C	6 A;10 N	0.20	56.1	16.7	20.0	A 1.25;N – 0.34
D	A	6 A;10 N	0.20	>0.001	X	X	X
D	B	6 A;10 N	0.20	>0.001	20.0	12.0	A 3.99;N – 1.29
D	C	6 A;10 N	0.20	>0.001	30.0	16.0	A 4.77;N – 0.83

Appendix

Two-way Statistical Analysis:

```
                    Class          Levels    Values
                    treatment           3    C D L
                    stage               2    Adult Juvenile
                                Number of observations    18
NOTE: Due to missing values, only 16 observations can be used in this
analysis.
Dependent Variable: mortality
                                        Sum of
Source                      DF          Squares      Mean Square    F Value    Pr > F
Model                        5     1501.782708       300.356542       2.92    0.0699
Error                       10     1026.966667       102.696667
Corrected Total             15     2528.749375

                  R-Square     Coeff Var      Root MSE    mortality Mean
                  0.593884      54.75953      10.13394          18.50625
Source                      DF        Type I SS     Mean Square    F Value    Pr > F
treatment                    2     202.9077083     101.4538542       0.99    0.4060
stage                        1     574.8006250     574.8006250       5.60    0.0396
treatment*stage              2     724.0743750     362.0371875       3.53    0.0694
```

Source	DF	Type III SS	Mean Square	F Value	Pr > F
treatment	2	202.9077083	101.4538542	0.99	0.4060
stage	1	343.2385714	343.2385714	3.34	0.0975
treatment*stage	2	724.0743750	362.0371875	3.53	0.0694

Tukey's Studentized Range (HSD) Test for mortality

NOTE: This test controls the Type I experimentwise error rate, but it generally has a higher Type II error rate than REGWQ.

Alpha	0.05
Error Degrees of Freedom	10
Error Mean Square	102.6967
Critical Value of Studentized Range	3.15106
Minimum Significant Difference	11.29

Means with the same letter are not significantly different.

Tukey Grouping	Mean	N	stage
A	24.500	8	Juvenile
B	12.513	8	Adult

Adjustment for Multiple Comparisons: Tukey-Kramer

treatment	stage	mortality LSMEAN	LSMEAN Number
C	Adult	5.5666667	1
C	Juvenile	22.6666667	2
D	Adult	25.0000000	3
D	Juvenile	14.0000000	4
L	Adult	11.1333333	5
L	Juvenile	33.3333333	6

Least Squares Means for effect treatment*stage
Pr > |t| for H0: LSMean(i)=LSMean(j)
Dependent Variable: mortality

i/j	1	2	3	4	5	6
1		0.3731	0.3578	0.9348	0.9812	0.0598
2	0.3731		0.9998	0.9276	0.730	0.7847
3	0.3578	0.9998		0.8766	0.6726	0.9377
4	0.9348	0.9276	0.8766		0.9995	0.3626
5	0.9812	0.7302	0.6726	0.9995		0.1629
6	0.0598	0.7847	0.9377	0.3626	0.1629	

According to my PR>F value for the overall experiment my results are moderately significant with a PR>F value of 0.070. The PR>F value for stage is significant at 0.0396. Interaction analysis indicated that adult mortalities are highest in the dark and juvenile mortalities are highest in the light and lowest in the dark. Analyzing treatment means shows that the mean mortalities for light and dark are opposite for juveniles and adults, supporting my hypothesis.